Appendix A

Aquatic Invasive Species That Threaten Utah

From Kent Hauk: I would like to see bullet-like preventions and controls specifically dealing with each AIS and for all user groups. For example, I think a list including each known AIS should be presented here or somewhere in the plan. Some control or prevention activity can be presented with each AIS and accomplished with minimal expense. This will show that the agencies involved in implementing the plan are seriously involved in natural resource stewardship and not just doing this to gather a paycheck. Governmental agencies should not only administer this program to the public, but they should also hold themselves accountable under the same standards. Here's an example. Whirling disease causative spores are spread in the mud by fishermen and vehicles. What does each agency commit to do to prevent its further spread? Along with the routine outreach to the fisherman and other public, each governmental agency involved with natural resources should commit to something as simple as requiring the washing of their vehicles after they have left an area endemic to whirling disease and before entering another area. If the public is required to decontaminate a boat after leaving a contaminated site, then the vehicle operated by the government sector should be cleaned after leaving a contaminated area also. "What's good for the goose.....".

A. Aquatic invasive species (AIS) are not strangers to Utah. In fact many species now inhabit Utah and others threaten the state with immediate arrival. The list frequently grows with discoveries of new species or new threats, and it includes pathogens (2), fungi (1), algae (1), plants (3), mollusks (11), fish (3), amphibians (4), and reptiles (2). Their biographic accounts follow and the accounts are arranged in phylogenetic order.

Aquascaping (Crystal Stock--done)

Aquarium dumping (Dan Keller--done)

Bait Releases (E.Freeman--done)

Pathogens

Whirling Disease (C.Wilson or designee)

Viral Hemorrhagic Septicemia (C.Wilson or designee)

Private Aquaculture (T.Miles & L.Dalton)

Fungi and Algae

Reference authority ????? (intro L.Dalton)

Chytrid fungus (*Batrachochytrium dendrobatidis*) (E.Freeman)

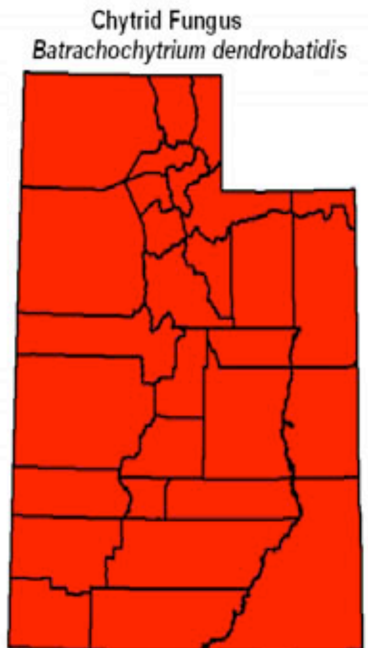
Ecology: Chytrid fungus is responsible for a deadly amphibian disease known as Chytridomycosis. The origin of this fungus is unknown. The spores of this fungus attack the keratin in frog skin. Due to a frog or toads ability to breath and drink through its skin, this attack of the skin makes it very difficult to perform these tasks. These fungal spores can also damage the nervous system of the victim, which affects the frog's behavior.

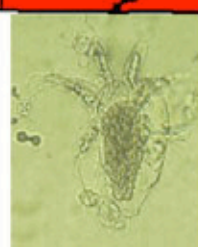
There are several signs to look for when trying to determine if you have an affected frog. They can have discoloration of the skin, usually having a reddish hue. There can be peeling or sloughing on the outside layers of the skin. Another skin related symptom can be the frog's skin having a rough texture instead of being smooth to the touch. Infected individuals tend to be very sluggish with no perceived appetite. They also tend to sit out in the open, seemingly having no intent of protecting itself by hiding. Another characteristic of infected frogs is the lack of ability to hold their limbs close to their bodies. In extreme cases the frog's legs actually trail behind the body.

Distribution: This fungus is found worldwide. It is presently found in Australia, Africa, North, Central and South America, Europe, New Zealand and Oceania. It is presently found in various portions of the United States including Utah. The potential for the fungus to be found throughout the US is very high.

Pathways of Introduction: It is not known how Chytrid fungus came to the United States and spread so effectively. Museum specimens from Colorado and California show that the fungus has been here since at least the 1970's. There are several vectors that can spread the fungus. Humans are a major factor in the spread of this fungus. We can pick up the fungus unknowingly from an infested area and transport it to a new area if we do not decontaminate equipment. Migratory birds and other animals can also transport the spores to new sites after picking up the spores in infected waters. The frogs themselves act as a vector moving the spores to new waters as they travel throughout their range.

Management Considerations: There is no known way in which to eradicate Chytrid fungus from the wild. Decontamination of equipment is the best practice in helping to halt the spread of this fungus. Spraying down all equipment with 409 cleaner and then letting it dry in the sun effectively kills the spores. There are currently ongoing research projects working with anti-fungal agents, but there have been no definitive results at the current time.





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Rock Snot (*Didymosphenia geminata*) (D.Keller)

Photo by Sarah Spaulding, USGS and EPA

Didymo covers approximately 50 percent of the substrate in this image from Rock Creek, Utah.

Ecology**:** Rock Snot is a diatom, which is a type of single-celled algae. Diatoms are remarkable organisms, unique for their silica (SiO2) cell walls. Diatoms are found in nearly every freshwater and marine aquatic habitat and contribute a large percentage of the global carbon budget through photosynthesis. D. geminata is made up of cells that cannot be seen with the naked eye until large colonies form. It only needs one of these cells to be transported for the algae to spread (Biosecurity NZ, 2005). In both oceans and freshwaters, diatoms are one of the major groups of organisms within the plankton (including other algae, bacteria, and protozoa) and also grow attached to surfaces. The life history of diatoms includes both vegetative and sexual reproduction (reviewed in Edlund & Stoermer 1997), although the sexual stage has not been documented in D. geminata (but see Skabichevsky 1983). D. geminata cells possess a raphe, a structure that allows the cells to move on surfaces. The cells also have an apical porefield, through which a mucopolysaccaride stalk is secreted. The stalk may attach to rocks, plants, or any other submerged substrate. When the diatom cell divides (i.e. vegetative reproduction), the stalk also divides, eventually forming a dense mass of branching stalks. It is not the diatom cell itself that is responsible for the negative impacts of D. geminata, but the massive production of extracellular stalk. Extra cellular polymeric substances (EPS) that comprise the stalk are predominantly composed of polysaccarides and protein. They are complex, multi-layered structures that are resistant to degradation. The degree to which internal (genetic) and external (environmental) change initiates the high level of stalk production is unknown, yet resolving the mechanisms of stalk production is crucial for determining ecological impacts, physiological regulation, and control of D. geminata. Currently little is know of the biology and ecological roles of D. geminata, and we need basic information to determine the causes and conditions that lead to nuisance blooms and the geographic expansion of this diatom.

Distribution: Know locations in Utah include Cottonwood Gulch near Joes Valley Reservoir, and Rock Creek on the south slope of the Uinta Mountains. The enclosed map shows distribution within the United States.

Invasion pathways: The mechanisms for D. geminata to expand its range to new watersheds are not well understood. Early suggestions that increases in UV-B radiation was tied to the expansion were not supported (Sherbot & Bothwell 1993, Wellnitz et al. 1996, Rader & Belish 1997). Recent work illustrates the capacity of D. geminata to survive outside of the stream environment as well as potential vectors in its spread. Cells are able to survive and remain viable in cool, damp, dark conditions for at least 40 days (Kilroy 2005). Fishing equipment, boot tops, neoprene waders, and felt-soles in particular, all provide a site where cells remain viable, at least during short-term studies (Kilroy et al. 2006). At the same time, prime destinations for fishing are becoming more popular with anglers. Rather than frequent a favorite local fishing site, it is now common that anglers travel to multiple, or distant destinations for fishing vacations. Moreover, they may be fishing in a river less than twenty-four hours after leaving their local rivers in North America, and unknowingly spreading D. geminata.

The arrival of D. geminata in New Zealand in 2004 indicates that it most likely arrived via human-assisted means, for example on footwear, fishing equipment, boats, etc. (Kilroy, 2004).

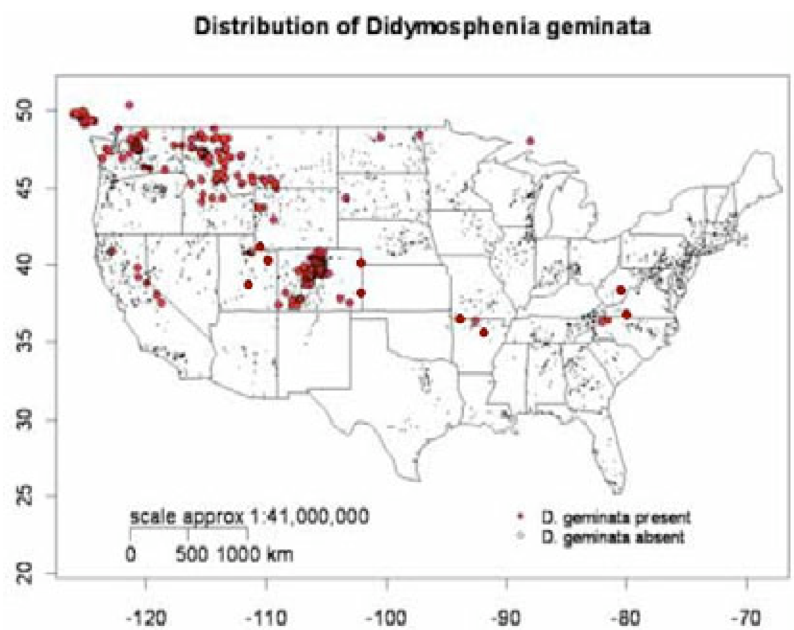
It is possible that clumps of D. geminata could pass through the guts of birds or other animals, or on the feet or feathers/fur of birds and animals (Atkinson, 1980; Kociolek and Spaulding, 2000; in Kilroy, 2004). Wind dispersal of mucilaginous material (the stalks) of D. geminata could occur over short distances (Kilroy, 2004).

Management Considerations: New Zealand is currently pursuing a series of experimental trials to test biocides for possible control of D. geminata within streams and rivers in New Zealand (Jellyman et al. 2006). In order to test the effectiveness of various biocides, D. geminata was grown on artificial substrates and placed in experimental stream channels. Numerous biocides were tested on D. geminata. The mats were exposed to each biocide for a period of one hour and the viability of algal cells determined at various time periods, up to 28 days after treatment. Mortality of fish in the experimental stream channels was also assessed. Of the five biocides tested, chelated copper had the greatest negative effect on D. geminata for all contact times. In the next stages, the tolerance limits of fish to chelated copper will be established. Although copper compounds have a long history of use as algaecides the United States, in lakes, reservoirs, and to a lesser extent, flowing waters, they have not been evaluated for control of D. geminata outside of New Zealand.

While D. geminata is not considered invasive in the United States, the diatom's nuisance blooms has economic impacts. The human population of western United States is closely dependent on a system of canals to transport water for hydropower generation, agriculture, and human consumption. Nuisance algae, including D. geminata, regularly thrive on the stable substrate and flow regime of canal systems (Pryfogle et al. 1997). In some canal systems, managers implement regular removals by scraping D. geminata growths from the concrete surfaces of canals.

Didymosphenia geminata is often reported by recreationalists to land managers as being unsightly. The stalks are often mistaken for raw sewage, leading homeowners and recreationalists to complain to local water treatment plants. Many communities rely on tourism dollars that are generated by outdoor recreation. Natural resource opportunities represent important economic value, yet they may be vulnerable to damage by the spread of this nuisance species.

An aggressive education and outreach program is required to change water resource user behavior in order to minimize spread of D. geminata on a global scale.
A public awareness campaign, directed at freshwater anglers, boaters, professional guides, and other recreationalists must be integrated with existing invasive species programs. Freshwater resource users, including ecologists, water managers, fisheries biologists, and other scientists, need to be aware of the threat and should practice decontamination procedures to prevent the spread.



Aquatic Plants

Reference authority ???, also see Mr. Steve Dewey, USU; he is the author for Weeds of the West and may have online database (intro J.Polloczek)

Common Reed (*Phragmites australis)* (J.Polloczek)

Ecology: Phragmites is a tall, perennial, sod forming grass or reed (Uchytil 1992; Amsberry et al. 2000). Long pointed leaves grow from thick vertical stalks and flowers form dense clusters that create a plume-like flower head tawny in color (ISSG 2006). *Phragmites* forms dense monodominant stands along marshes and shorelines (Uchytil 1992). These dense stands of tall reeds crowd native plants, displace native wetland vegetation and alter nutrient cycling (Saltonstall 2002;Windham and Ehrenfeld 2003). These changes alter the structure and function of some marshes and can threaten wildlife populations (Roman et al. 1984).

The common reed reproduces both by seed and vegetative means. Seeds are dispersed by wind and water and can persist in the marsh following a draw down as part of the seed bank. Most reproduction, however, is vegetative through the use of an extensive network of rhizomes and stolons (Smith and Kadlec 1983).

Distribution: Phragmites is native to North America and found in every U.S. state (U.S. Army Corps of Engineers 2004). The rapid increase of *Phragmites* in North American wetlands, however, is due to colonization by a more aggressive European variant of the plant (Saltonstall 2002). *Phragmities* is now common to wetland areas and canals throughout most of Utah (USDA, NRCS 2008).

Pathway of Introduction: Once established, *Phragmites* spreads rapidly by means of rhizomes or stolons (Uchytil 1992). *Phragmites* can spread up to 15 or 20 feet per year from vegetative spread alone. The flooding of the Great Salt Lake in the 1980's is believed to be an important factor in the dramatic increase of *Phragmites* around the eastern shore of the Great Salt Lake (personal communication with Val Bachman, Area Waterfowl Manager). Increased physical disturbances in marshes can initiate and accelerate expansion such as disturbances by foot traffic and floating debris (Amsberry et al. 2000).

Management Considerations: Currently there are 26 herbivores in North America known to attack *P. australis* (Tewksbury et al., 2002). Only five of these herbivores are believed to be native. Within this group only the Yuma skipper, *Ochlodes yuma*, a dolichopodid fly in the genus *Thrypticus*; and a gall midge, *Calamomyia phragmites*, are considered native and monophagous on *P. australis* (Tewksbury et al. 2002). Possible biocontrol species are being tested, but are not currently available (Blossey 2003).

Only mechanical and chemical control methods are available at this time for management of *Phragmites*. Mechanical control includes plowing, crushing, mowing, dredging and burning. Mechanical control methods that break up plant matter should be used with caution as they have the potential to increase vegetative spread. Prescribed burning can be successful only if root burn occurs. Burning is recommended during the summer when carbohydrate reserves in the plant are low and when the soil is dry for maximum root burn (Uchytil 1992). Burning removes accumulated *Phragmites* leaf litter, allowing the seeds of other species adequate area to germinate (Marks et al. 1993). Complete removal of *Phragmites* by burning alone, however, is difficult and the practice is typically coupled with herbicide treatment and/or water draw downs.

The U.S. Army Corps of Engineers suggests a glyphosphate such as Rodeo® or Imazapyr, Arsenal® as possible herbicide control. Rodeo® should be applied during late summer or fall when plants are actively growing and in full bloom. Arsenal® is nonselective and will kill other desirable plants. The 2, 4-D herbicides (SEE 2, 4-D, Weed Rhap A-6D, and Weedar 64) are also registered for use on canals or ditch banks in Utah (U.S. Army Corps of Engineers 2004). The Division of Wildlife Resources is actively using a combination of glyphosphate herbicides and prescribed burning to control *Phragmites* along the eastern shore of the Great Salt Lake.

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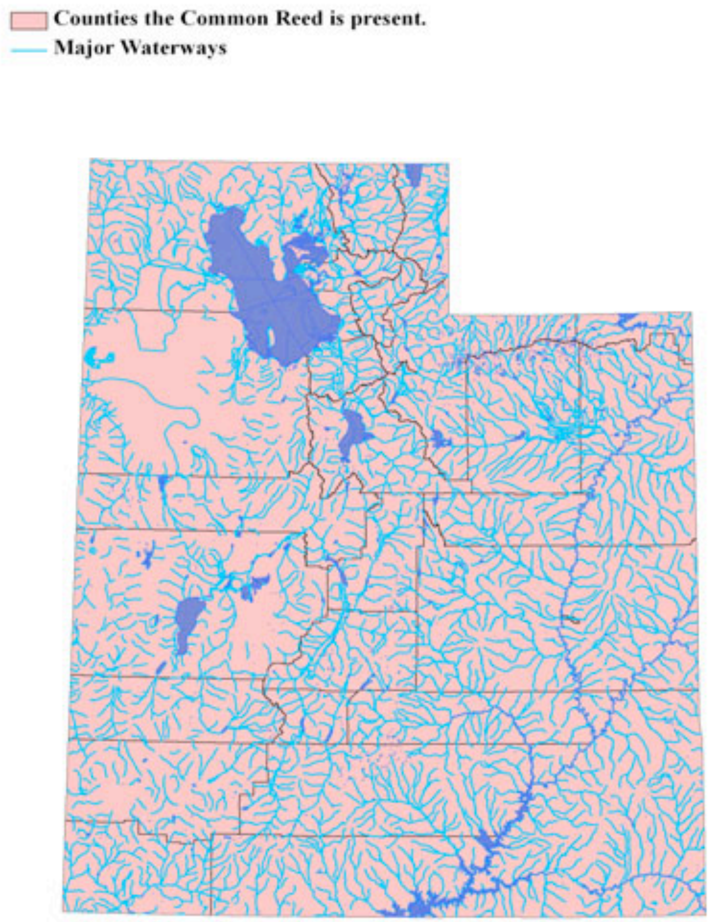
Common Reed



Richard Old
XID Services, Inc.,
Bugwood.org

Tamarisk (J.Polloczek)

Purple loosestrife *(Lythrum salicaria)* (J.Polloczek)

Ecology: Purple Loosestrife is an emergent, rhizomatous, perennial with erect stems. The leaves are simple, entire and opposite or whorled with rose-purple flowers consisting of 5 to 7 petals (Whitson et al. 1996). Purple loosestrife prefers aquatic sites along stream banks and shallow ponds, though it has successfully invaded drier regions by utilizing irrigation canals and waterways as pathways to dispersal (Whitson et al. 1996). *L. salicaria* prefers moist soils of neutral to slightly acid pH, however it is found in a wide range of soil textures and types and is able to adjust to seasonal or semi-permanent changes in water levels (Thompson et al. 1999).

The successful spread of purple loosestrife is attributed to its ability to reproduce through seed or vegetative means, prolific seed production and a wide scope of dispersal mechanisms. A mature plant can produce up to 2.7 million seeds and disturbance to underground stems increases spread by encouraging new growth from adventitious shoots and roots (Thompson et al. 1999).

Purple loosestrife has drastically altered wetlands across North America (Thompson et al. 1999). Once *L. salicaria* is established, it outcompetes and replaces native plants (Gaudet and Keddy 1995) that provide higher quality food and habitat for wildlife (Raloff 1992; Brown et al. 2002). *L. salicaria* forms dense homogeneous stands that restrict native wetland plant species and reduce future reproduction by native plants through competition for pollinators (Thompson 1987; Brownet al. 2002). The recreational and overall aesthetic value of wetlands and waterways is diminished as dense stands of *L. salicaria* choke waterways and decrease biodiversity

Distribution*:* Purple Loosestrife is of Eurasian origin and has been established in North America since the early 1800's. This species has expanded its distribution from its point of introduction in the northeast to the western U.S. and north into Canada (Thompson et al. 1999). Purple loosestrife currently inhabits 43 of the 48 contiguous states and is prevalent in Utah's northern wetland areas (Sturtevant 2008).

Pathways of Introduction: Purple loosestrife spreads downstream through water dispersal of seeds and vegetative matter. Seeds are unintentionally transported and spread with wetland soil carried by animals, humans, boats and vehicles (Thompson et al. 1999). Purple loosestrife is also widely sold as an ornamental in states where regulations do not prohibit its sale and distribution. In Utah, purple loosestrife is listed as a noxious weed and its sale is prohibited.

Management considerations: The best control measure, as with many invasive plants, is to preserve a healthy native ecosystem to prevent or slow invasion (ISSG 2006).

Herbicides are the most commonly used method of control for purple loosestrife. Commonly used chemicals include glyphosphate sold as Rodeo® for use in wetlands and Roundup® for use in uplands, 2, 4-D, and Renovate®. However, glyphosphate is nonselective and can kill desirable plants associated with loosestrife if applied carelessly (Butterfield et al. 1996). Multiple chemical treatments are usually required for control as new seedlings emerge annually from the seed bank.

Biological control methods are more effective for long-term control of larger populations of purple loosestrife. In North America four insects have been approved by the U. S. Department of Agriculture for use as biological control agents: the root-mining weevil *Hylobius transversovittatus*, two leaf-feeding beetles *Galerucella calmariensis* and *G. pusilla*, and *Nanophyes marmoratus*, a herbivorous weevil. The impact of these introduced beetles on native, non-target species is considered low. *G. calmariensis* has provided successful control of purple loosestrife (Malecki and Blossey 1993).

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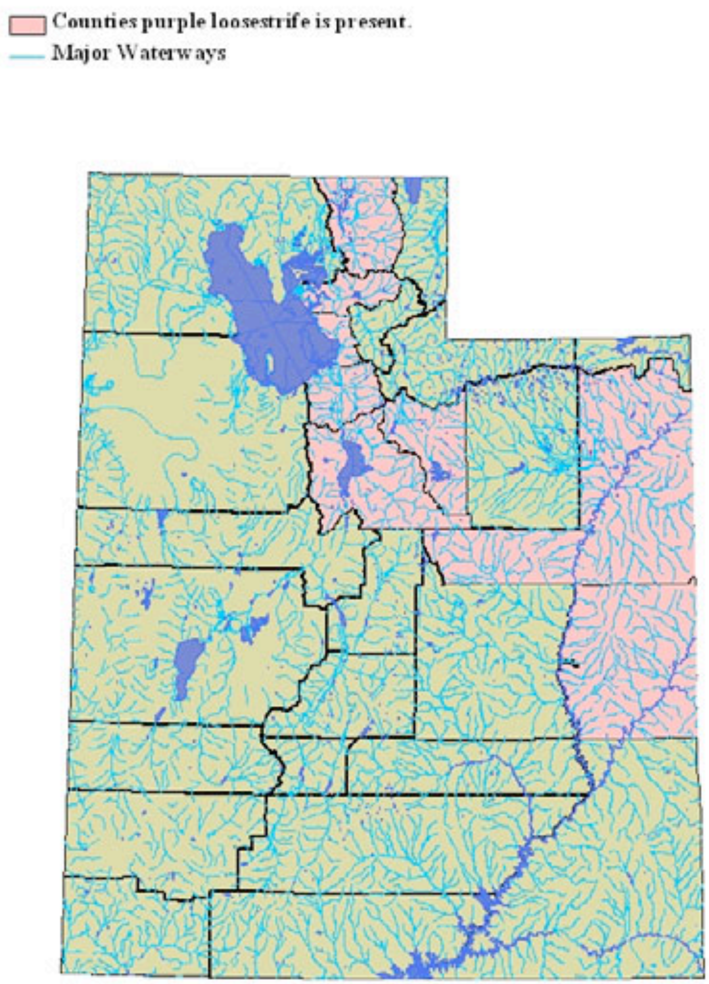
Purple Loosestrife



Paul Champion, NIWA

Eurasian Water Milfoil (J.Polloczek) …………………………………….

Curly Pond Weed (J.Polloczek) ………………………………………….

Invertebrates

Reference Utah Comprehensive Wildlife Conservation Strategy (Wildlife Action Plan) noting that non-native fish species compete with either Tier I (T&E), Tier II (species of conservation concern) or Tier III (species with at-risk habitats) native species and cite (1) Utah Wildlife Code, and (2) Collection, Importation & Possession of Zoological Animals as authorities. (intro L.Dalton)

Mollusks

New Zealand mudsnail (*Potamopyrgus antipodarum*) **(J.Polloczek)**

Ecology: *P. antipodarum* is a small (<5mm) invasive, hydrobiid snail. It has an elongate, dextral shell that varies in color and typically has 5 to 6 whorls at maturity (Gustafson 2005). New Zealand mudsnails (NZMS) are able to invade and grow in a wide range of ecological habitats. They are found in rivers, reservoirs, lakes, and estuaries and are able to adapt to a wide range of temperature, salinities and substrates (Zaranko et al.1997; Richards et al. 2001; Hall et al. 2003). NZMS are not able to withstand freezing temperatures at any salinity (Hylleberg and Siegismund 1987). The highest densities of NZMS typically occur in systems with high primary productivity, constant temperatures and constant flow (Gustafson 2005).

Reproductive, behavioral and morphological adaptations have made NZMS an ideal aggressive invasive species. Their rapid spread is attributed to high reproductive and growth rates, parthenogenesis and lack of parental care. A single female can theoretically produce up to 3.125 x108 snails in one year. The ability for this species to reproduce asexually means that it is possible for a single individual to produce a new population (Zaranko et al. 1997). The presence of an operculum also allows them to survive for several weeks out of water (Bowler 1991).

NZMS are shown to negatively impact the aquatic communities they invade. Hall et al. (2003) found NZMS population densities that exceeded 100,000 individuals per square meter and consumed 75% of the gross primary production. NZMS outcompete native invertebrates for food and space and have also been shown to contribute to weight loss in fish when consumed (Bowler 1991; Vinson and Baker 2007). There is also concern that the high densities of NZMS could produce biofouling in facilities that become infested (Zaranko et al. 1997).

Distribution: *P. antipodarum* has spread from New Zealand to freshwater environments throughout the world. This species current distribution includes: Australia, Europe, Asia, and North America. First discovered in the United States in 1987 in the Snake River near Hagerman, Idaho; NZMS are now locally abundant in western rivers (Bowler 1991; Dybdahl and Kane 2005). In Utah, NZMS are found in most of the major river drainages of the northern part of the state and in the Green River (Gustafson 2005; Harju 2007).

Pathways of Introduction: The original source of introduction is unknown, though it is speculated that NZMS was introduced through the commercial transport of aquaculture products (Bowler 1991). Since introduction, both active and passive transport methods have contributed to its spread. NZMS have been shown to spread independently upstream through locomotion. Passive spread by birds, through the alimentary canal of fish, and contaminated recreational equipment is also documented (Haynes et al. 1985; Richards et al. 2004; New Zealand Mudsnail Management and Control Plan Working Group 2006).

Management considerations: Spread of NZMS can be prevented through increased public education efforts. NZMS have no resistant stage or adhesive structures like other aquatic nuisance species and simple preventative measures can reduce their likelihood of spread to new areas. Once established, however, NZMS are extremely difficult to remove. The spread of NZMS into new watersheds is primarily through unintentional human transport on contaminated recreational equipment, water containers and bait buckets. (Richards 2002). Desiccation and freezing may be used to decontaminate angling and other recreational equipment that comes in contact with water, but this method can be slow, taking up to 24 hours. A faster (less than 30 minutes) and more effective alternative is to spray or immerse gear in disinfectant baths of: copper sulfate, benzethonium chloride, Formula 409® or Sparquat® (Hosea and Finlayson 2005; New Zealand Mudsnail Management and Control Plan Working Group 2006).

Possible control methods of existing populations include periodic: molluscicide application, desiccation of the waterbody, and introduction of a biological control agent. GreenClean® is a non-copper-based algaecide that has been successful at killing NZMS in lab experiments and is being tested for field applications (New Zealand Mudsnail Management and Control Plan Working Group 2006). Biocontrol lab trials using a trematode parasite from the native range of New Zealand mudsnails have been positive so far (Dybdahl et al. 2005), though this method of control is currently unavailable.

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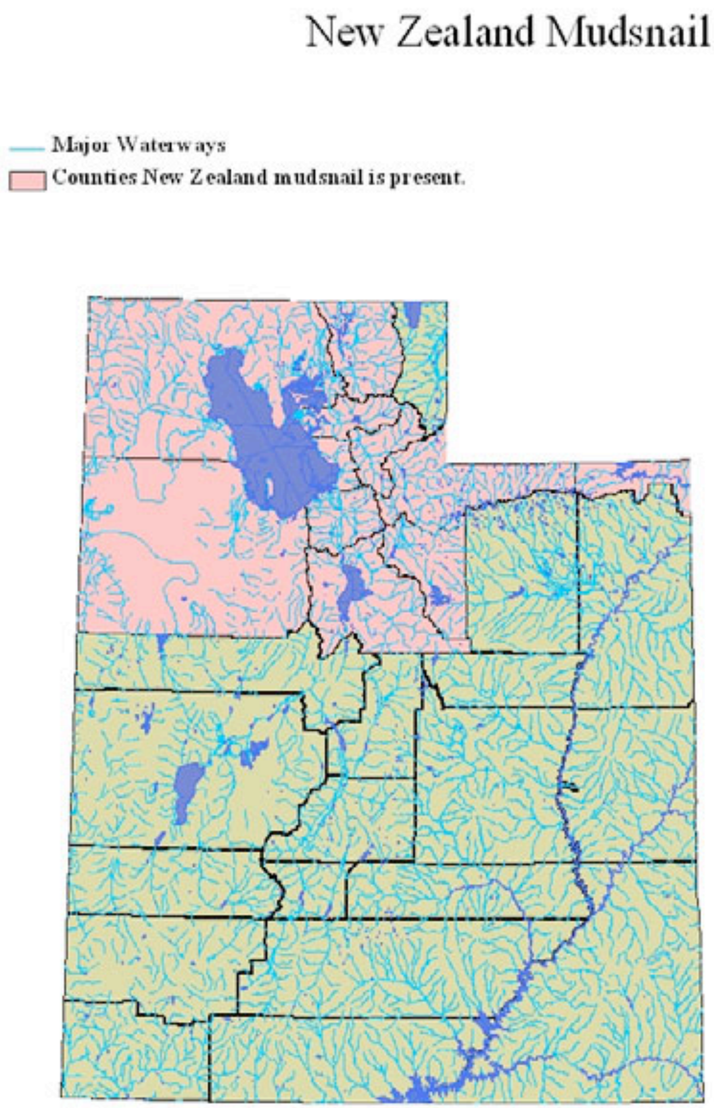

Daniel L. Gustafson
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Red-rimmed Melania (*Melanoides tuberculatus*) (E.Freeman)

Ecology: This is a small, aquatic, herbivorous snail, consuming detritus and benthic microalgae. Adult snails typically attain a shell length of between 30 and 36mm, however there have been reports of snails achieving lengths up to 80mm. It has an elongated conical shell with regularly increasing whorls. Five whorls typically make up the shell. There are prominent vertical ribs present on the middle and upper whorls. The spiral of the shell is usually twice the length of the aperture or more. Shell coloration is usually light brown, frequently mottle with rust colored spots that may for a spiral below the suture.

Red-rimmed Melania is very common throughout its native range in both Africa and Asia. It prefers shallow, slow running water (0.6 – 1.2 cfs). This snail tolerates a wide range of saline environments and can be found in fresh water as well as estuarine environments up to 30 ppt. The temperature tolerance for this snail is believed to be restricted in the US to between 18 – 25 degrees Celsius. The prime habitat for this species consists of areas rich in detritus and silt behind overhanging stems and protruding roots of bank vegetation. They are active mostly at night, hiding beneath decaying plants and stones or burying themselves in the mud during the day.

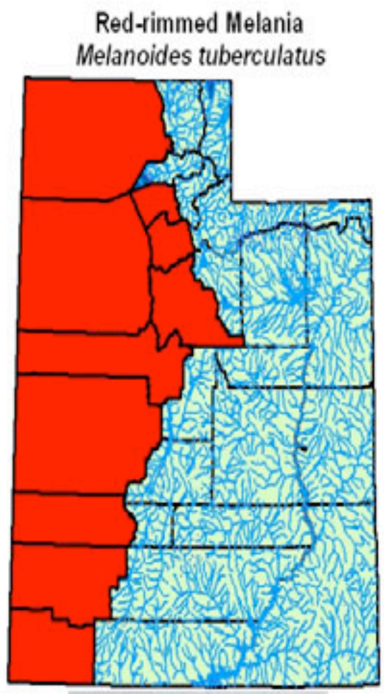
Red–rimmed Melania reproduce both sexually and through parthenogenesis. Individual snails as small as 10mm are able to reproduce. This species is viviparous having up to 70 offspring held in a brood pouch. They remain in the brood pouch until released at 1 – 2mm in length.

Red-rimmed Melania is also a vector for several important diseases. They are intermediate host for a number of trematode parasites including: *Clonorchis sinensis*, the Chinese liver fluke; *Paragonimus westermani*, the Oriental lung fluke; *Diorchitrema formosanum*, and intestinal trematode; *Opisthorchis sinensis*, the human liver fluke; and *Philophthalmus sp*., the avian eye fluke.

<u>Distribution</u>: Native to subtropical and tropical regions of northern and eastern African and southern Asia from Morocco and Madagascar to Saudi Arabia, Iran, Pakistan, India, southern china, and Indonesia east to Java and Celebes. In the United States it is widely distributed throughout the Gulf of Mexico ecosystem. A San Francisco aquarium dealer prior to 1937 introduced it into California. It was then introduced into Tampa Bay, Florida after purchase from the same San Francisco aquarium dealer. There are a number of springs throughout the Great Basin that either have Red-rimmed Melania or are suitable for their survival.

<u>Pathways of Introduction</u>: The original method of introduction to the United States was through the aquarium trade. It is likely that this is how it was spread to the Great Basin, including Utah. Fisherman using felt-soled waders as they move from one site to the next without decontaminating their equipment can move it throughout Utah

<u>Management Consideration</u>: Once these snails have been introduced into a new body of water it is unlikely to remove them. The best method for preventing the spread of this species into new waters is to decontaminate all equipment that has come in contact with infested waters. This can be done with scalding hot water or an easier method of spraying equipment down with 409 cleaner and letting the equipment dry in the sun.



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Chinese mysterysnail (E.Freeman)

Asian Clam (*Corbicula fluminea*) (D.Keller)

Description: The outside of the shell is normally yellow-brown with concentric rings. The color can flake, leaving white spots. The inside of the shells are pearl to purple in color. Although the Asian clam grows and disperses less rapidly than the zebra mussel, it too is causing considerable fouling problems and is threatening native species. Costs associated with its fouling damage are about $1 billion/yr (Isom 1986; OTA 1993).

Ecology: Asian clams are bi-valve filter feeders that remove particles (plankton) from the water column. They can be found at the sediment surface or slightly buried. The ability to reproduce rapidly coupled with low tolerance of cold temperatures can produce wild swings in population sizes from year to year in northern water bodies. *C. fluminea* is found both in lotic and lentic habitats over its native range in southeastern Asia. In the United States it has been most successful in well-oxygenated clear waters (Belanger et al., 1985; Stites et al., 1995). Maximum Asian clam density has been reported to vary between 1000/m2 (Gotfried and Osborne, 1982; Stites et al., 1995) to 6000/ft2 (Sinclair, 1971a) and even 25,000/ft2 (Sinclair, 1971b). Life span varies according to habitat, with a maximum life span of approximately 7 years (Hall, 1984). They can self fertilize and release up to 2,000 juveniles per day, and more than 100,000 in a lifetime. Juveniles are only 1mm long when discharged and take one to four years to reach maturity. At this time they are about one centimeter long. Adults can reach a length of about 5 cm. Usually *C. fluminea* is more common and occurs at higher densities in stream pools than in stream runs (Blalock and Herod, 1999). Fine clean sand, clay, and coarse sand are preferred substrates, although they may be found in lower numbers on most any substrate (Gottfried, and Osborne, 1982; Belanger et al., 1985; Blalock and Herod, 1999).

Asian clams can tolerate salinities of up to 13ppt for short periods of time. If allowed to acclimate, they may tolerate salinities as high as 24ppt (King et al., 1986). Optimum is at lower salinities (Morton and Tong, 1985). In nature, Asian clams occur mostly in freshwaters, however, they have been reported from brackish and estuarine habitats, but are typically not as abundant in such habitats as in freshwaters (Carlton, 1992).

This species appears to tolerate low temperatures well. Viable populations have been reported surviving temperatures of 0-2°C over winter in the Clinton River, Michigan (Janech and Hunter, 1984). However, low temperatures do limit reproduction, since veligers are typically released at temperatures of 16°C or higher (Hall, 1984).

Distribution: The first collection of C. fluminea in the United States was recorded in 1938 along the banks of the Columbia River near Knappton, Washington (Counts 1986).

Currently it is found in 38 states and the District of Columbia. In Utah, Asian clams have been established in Lake Powell since the mid 1970’s. It is likely they were present in the Colorado River prior to completion of the Glen Canyon Dam in 1960. Recently they have been found at various locations along the Jordan River, which flows from Utah Lake, into the Great Salt Lake. The Jordan River provides water to a significant canal system, so the clams are likely all over Utah Valley and the Salt Lake Valley, which is where most of Utah's 2.5 million people live. Utah Lake is an essential element of the Central Utah Project, receiving water as a trans-basin diversion from the Colorado River drainage via Strawberry Reservoir. The reservoir receives water from 10 south slope Uinta Mountain drainages via an extensive underground collection system. Those drainages would have eventually entered the Green River and the Colorado River, which drain to Lake Powell. The fouling effects of Asian clams will likely create problems within this system. *C. fluminea* was confirmed in Willard Bay (both its inflow and outflow) in the Spring of 2007; it receives water from the Weber River. This species is also found in Yuba Reservoir in south central Utah. (See figure 1 for Utah distribution.)

Pathways of Introduction: *C. fluminea* was thought to have first entered the United States as a food item. Humans are the primary agent of dispersal. They are thought to spread primarily through activity such as bait bucket introductions, accidental introductions associated with imported aquaculture species, and intentional introductions by people who buy or sell them as a food item in markets. The only other significant dispersal agents are water currents or flooding events.

Management Considerations: *C. fluminea* populations are controlled by a variety of methods. Where intakes pipes are fouled, thermal regulation is employed, whereby water in the pipes is heated to temperatures exceeding 37 degrees Celsius. But this method is not feasible in most existing water systems. Mechanical methods, such as using screens and traps, can effectively dispose of older clams and remove body tissue and shells from the system. Chemicals, such as small concentrations of chlorine or bromine, are used to kill juveniles and sometimes adults. This method is very effective, but because of increasing restrictions on the amounts of these chemicals that may be released from a facility, managers have been moving away from this method.

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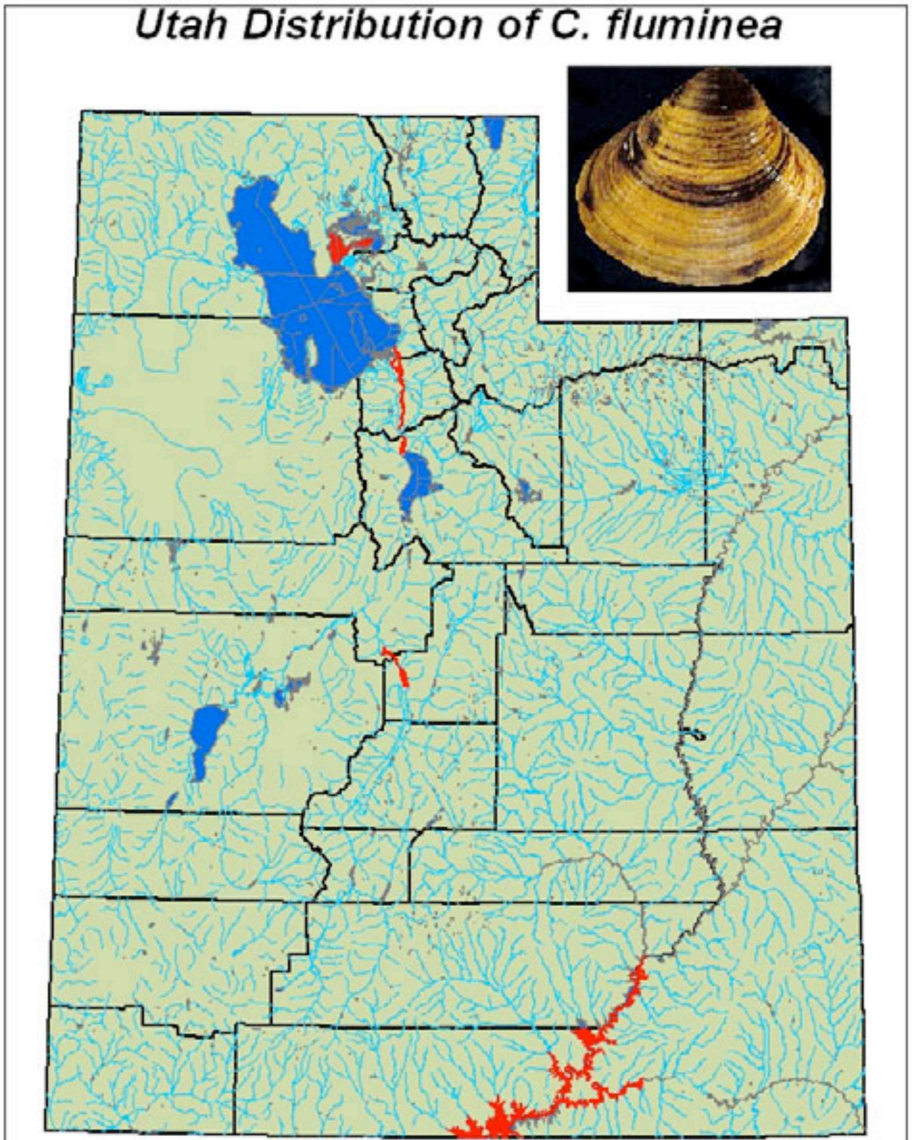


Figure 1

Crayfish (D.Keller)

Utah has three know species of invasive crayfish. These species are the northern crayfish (*Orconectes virilis*), Louisiana/red swamp crayfish (*Procambarus clarkii*) and the pacific crayfish (*Pacifasacus leniusculus*). Another species of concern is the water nymph crayfish (*Orconectes nais*). This crayfish is currently not found in Utah; however, it has heavily infested Colorado waters. Due to its distribution on the western slope of Colorado it is likely that it will invade Utah waters. Rusty crayfish, (*Orconectes rusticus*) is also not found in Utah but poses a threat due to its wide North American distribution and its popularity among anglers as bait. Environmental impacts of crayfish introductions can be positive, negative or neutral. However, non-native crayfish introductions have shown the potential to negatively impact ecosystems and cause economic damage. Negative effects of non-native crayfish introductions include displacement of native crayfish species, transfer of disease, consumption of fish eggs, reduction of fish stocks, consumption of large amounts of macrophytes, indirect and direct effects on other invertebrates and destabilizing ditches, canals, and stream banks. Utah has one know native crayfish, (*Pacifastacus gambelii*); it is likely that any non-native crayfish introduction would place this species at risk. Law enforcement designed to prevent the spread of crayfish has proven difficult (many people intentionally spread crayfish to enhance recreational sport/cray-fishing). The best method of control is to prevent their introduction. Educating anglers, crayfish trappers, bait dealers, and teachers about the threats posed by invasive crayfish will help reduce the risk of spreading.

Northern crayfish **(*Orconectes virilis*)**

Photo Credit: Keith A. Crandall

Description: According to Collicut (1998), *O. virilis* grows to a length of about 10-12 cm, not including the 2 pairs of long antennae or the large chelipeds (the large pincer bearing legs) that extend forward. Chelipeds often have a bluish tint, particularly in males, which have larger chelipeds and pincers than females. The head and thorax are covered by a shell-like carapace that is usually brownish to rusty red in color. They are found in permanent bodies of water deep enough not to freeze solid or experience low oxygen levels. *O. virilis* requires shelter in the form of rocks, logs, or thick vegetation in which to hide from predators during daylight hours.

Ecology: *O. virilis* eats some aquatic plants as well as invertebrates, such as snails and insects; it also eats tadpoles and small fish. They are probably best described as opportunistic omnivores consuming whatever they can catch. While they can catch some quick moving prey like tadpoles or fish, they probably obtain most of their food by scavenging dead animals.
O. virilis can mate in autumn or in spring. However, the eggs are not fertilized and laid

until spring. Females can store sperm from a fall mating and protect their eggs by carrying them under their tales. Eggs are attached to swimmerets in a large ball resembling a raspberry. The eggs hatch one to two months after they are laid. Young hatchlings look like miniature adults and can probably grow to about 2-3 cm long by the fall. *O. virilis* has a short lifespan. Males usually die after mating when they are about 2 years old. The females die after their young hatch, also at about 2 years of age. *O. virilis* occasionally lives longer, but it's thought that none survive beyond their 4th spring. (Collicut 1998)

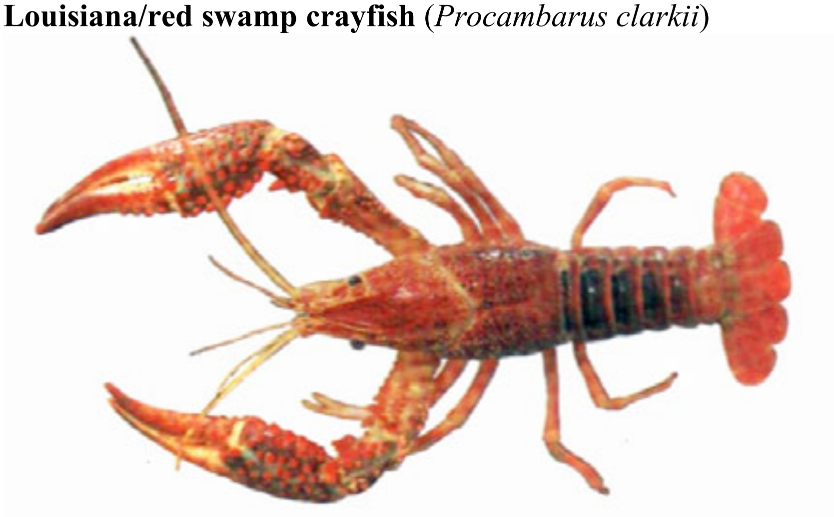
Distribution: (figure 1,Utah distribution).

Invasion pathways to new locations: Aquaculture: Crayfish are harvested from natural waters by commercial fishers and anglers or cultivated in small earthen ponds (Huner, 1997).
Live food trade: Crayfishes have been most commonly used as food and fish bait but are also commercially exploited in the pet trade as pets and food for predaceous pet fishes.

Management information: *O. virilis* is of serious concern because its burrows in ditches and levee banks may disrupt irrigation networks. *O. virilis*' burrowing and swimming activities may also muddy the water, reducing photosynthesis in submerged plants. (Godfrey, 2002)

Literature Cited:

Louisiana/red swamp crayfish (*Procambarus clarkii*)



Description: Usually colored a dark red, P. clarkii is capable of reaching sizes in excess of 50g in 3-5 months Adults reach about 5.5 to 12 cms (2.2 to 4.7 inches) in length.

Ecology: Unlike the native crayfish species of Europe P. clarkii is able to tolerate dry periods of up to four months (Henttonen and Huner, 1999; Ackefors, 1999). Because of this, it is able to occupy a wide variety of habitats, including subterranean situations, wet meadows, seasonally flooded swamps and marshes, and permanent lakes and streams. It thrives in warm, shallow wetland ecosystems. It can even be found in sluggish streams and lentic situations, being tolerant of low oxygen levels and high temperatures. It is one of few North American crayfishes with tolerance for saline waters (NatureServe, 2003).

In laboratory conditions *P. clarkii* preferred macro invertebrates to plant matter, preying largely on species with slow escape reactions (such as *Odonata, Ephemeroptera* and snails) and less on species with fast escape reactions, such as live mosquito fish (*Gambusia affinis*). Crayfish may be cannibalistic or prey on individuals of other crayfish species. P. clarkii prefers high-protein food (such as freshwater macro invertebrates) because it stimulates a high growth rate but is an opportunistic feeder and will consume plant matter and detritus when it is prey is lacking or it is unable to catch prey (Ilhéu and Bernardo, 1993, in Nystrom, 1999).

P. clarkii matures when it reaches a size of between 6 and 12.5 centimeters. A 10 cm female may produce up to 500 eggs, while smaller females may produce around a 100 eggs. The eggs are 0.4mm, notably smaller than those produced by members of the family Astacidae. Newly hatched crayfish remain with their mother in the burrow for up to eight weeks and undergo two moults before they can fend for themselves (Ackefors, 1999). Unlike the European native Astacus and Austropotamobius species, populations of *P. clarkii* contain individuals that are incubating eggs or carrying young throughout the year (Huner and Barr, 1994, in Lindqvist and Huner, 1999). This allows P. clarkii to reproduce at the first available opportunity, which contributes to its colonization success (Huner, 1992, 1995, in Gutierrez-Yurrita and Montes, 1999). In places with a long flooding period (greater than 6 months), there may be at least two reproductive periods (in autumn and spring). The spring period is longer and more prolific and persists until the drying of the marsh. For large females to reproduce it is necessary to have hormonal induction (produced by the photoperiod), a hydroperiod longer than four months, a temperature above 18 °C, and a pH between 7 and 8 (Gutierrez-Yurrita, 1997). If females have only a short period to prepare themselves for reproduction they must prematurely their burrow to feed; in such circumstances many females will die of dehydration, bringing about a depression in the population (Huner, 1995; Gutierrez- Yurrita, 1997, in Gutierrez-Yurrita and Montes, 1999).

P. clarkii exhibits a cyclic dimorphism of sexually active and inactive periods alternating during the lifecycle. After the young hatch, metamorphosis takes place, followed by two to three weeks of voracious eating. After this they moult and again assume their immature appearance (Hunter and Barr, 1994, in Ackefors, 1999). Egg production can be completed within six weeks, incubation and maternal attachment within three weeks and maturation within eight weeks. Optimal temperatures are 21-27 degrees and growth inhibition occurs at temperatures below 12 degrees Celsius (Ackefors, 1999). *P. clarkii* shows two patterns of activity, a wandering phase, without any daily periodicity, characterized by short peaks of high speed of locomotion, and a longer stationary phase, during which crayfish hide in the burrows by day, emerging only at dusk to forage. Other behaviors, such as fighting or mating, take place at nighttime. During the wandering phase, breeding males move up to 17 km in four days and cover a wide area. This intensive activity helps dispersion in this species (Gherardi and Barbaresi, 2000).

Distribution: (Figure 1, Utah distribution; Figure 2, North American distribution)
Native range: Northeastern Mexico and the south central USA (Henttonen and Huner, 1999).
Known introduced range: inter-state introductions into at least 15 other states in the USA

Invasion pathways to new locations: Agriculture: *P. clarkii* is a popular dining delicacy, accounting for the vast majority of crayfish commercially produced in the United States (Washington Department of Fish and Wildlife, 2003). It was the most dominant freshwater crayfish in the world during the 20th century and its commercial success led to intentional introductions throughout Spain, France and Italy during the 1970s and 1980s (Henttonen and Huner, 1999).

Natural dispersal: Natural dispersal from Spanish waters are thought to have facilitated the spread of *P. clarkii* into southern Portugal (Henttonen and Huner, 1999).
Other: *P. clarkii* can spread to new areas by anglers using them as bait. Popular as a bait species for largemouth bass, this is believed to have been the most likely cause for their introduction into Washington (The Washington Department of Fish and Wildlife, 2003).

Pet/aquarium trade: The habit of selling *P. clarkii* alive as an aquarium or garden pond pet may have accelerated the spread of the species through natural waterways in Europe (Henttonen and Huner, 1999).

Smuggling: The crayfish that now occur in African freshwaters are thought to have been introduced without the knowledge and permission of the relevant authorities (Mikkola, 1996, in Holdich, 1999).

Natural dispersal (local): There are reports of migrations of males over several miles in comparatively dry areas, especially in the rainy season (Nature Serve, 2003).
Other (local): P. clarkii can spread to new areas by anglers using them as bait (Aquatic Non-native Species Update, 2000).

Management considerations: When introduced into a suitable habitat *P. clarkii* may quickly become established and eventually become a keystone species (a primary contributor to the ecosystem it inhabits). Its introduction may cause dramatic changes to occur in native plant and animal communities (Schleifstein, 2003). For example, *P. clarkii* has contributed to the decline of native European crayfish (in the family Astacidae) by introducing interspecific competition pressure and acting as a vector for the transmission of the crayfish fungus plague, Aphanomyces astaci. *P. clarkii* has also been associated with the crayfish virus vibriosis in crayfish farms, and is an intermediate host for numerous helminth parasites of vertebrates (Thune et al., 1991; Hobbs III et al., 1989, in Holdich, 1999). *P. clarkii* also reduces the value of the freshwater habitats in which it occurs by consuming invertebrates and macrophytes and degrading river banks by its burrowing activity (Holdich, 1999).

Possible management options include the elimination (or reduction) of alien crayfish via

mechanical, physical, chemical or biological methods, the restocking of native crayfish populations (threatened by the crayfish plague fungus and interspecific competition with alien species), the development of plague-resistant strains of native crayfish and the use of legislation to prohibit the transport and release of alien crayfish.
Reduction may be possible by physical methods, although eradication is unlikely unless the population is particularly resitricted in range and size. All physical methods have environment costs, which should be weighed up against the environmental benefits of employing them. Mechanical methods to control crayfish include the use of traps, seine nets, and electro-fishing. Continued trapping is preferable to short-term intensive trapping, which may provoke feedback responses in the population such as stimulating a younger maturation age and a greater egg production. Physical methods of control include the drainage of ponds, the diversion of rivers and the construction of barriers (either physical or electrical).

Chemicals that can be used to control crayfish include biocides such as organophosphate, organochlorine, and pyrethroid insecticides; individual crayfish are differentially affected depending on their size, with smaller individuals being more susceptible. Since no biocides are crayfish-specific other invertebrates, such as arthropods, may be eliminated along with crayfish, and may subsequently have to be re-introduced. There is cause for concern about toxin bioaccumulation and biomagnification in the food chain (although this is less of a problem with pyrethroids). Another chemical solution lies in the potential to use crayfish-specific, or even species-specific, pheromones to trap animals. This has been used to control insect populations, but has not been researched with respect to crayfish, although crustaceans do use similar pheromones.
Possible biological control methods include the use of fish predators, disease-causing organisms (that infect crayfish) and use of microbes that produce toxins, for example, the bacterium Bacillus thuringiensis var. israeliensis (Pedigo, 1989, in Holdich et al., 1999). Only the use of predaceous fish has been used successfully; eels, burbot, perch and pike are predators are all partial to crayfish (Westman, 1991, in Holdich et al., 1999). The amount of cover, type of fish predator used and location are all important variables in determining the success of such an approach, and in general reduced coverage is correlated with increased predation rates.

Literature Cited:

Pacific crayfish (*Pacifastacus leniusculus*)
Also known as Californian crayfish or signal crayfish

Description: Its claws are robust and smooth on both surfaces, the underside is red in colour; with a single tubercle on the inner side of the fixed finger; and a white-turquoise patch on top of the junction of fixed and moveable fingers; adult males are massive either lengthways or in width. Males are up to 16 cm in length from tip of rostrum to end of telson, females up to 12 cm; much larger individuals have been recorded, i.e. 95 mm carapace length. The weight is typically 60 and 110 g at 50 and 70 mm carapace length. Its color is bluish-brown to reddish-brown, occasionally light- to dark-brown.

Ecology: *P. leniusculus* occupies a wide range of habitats from small streams to large rivers (e.g. Columbia River) and natural lakes, including sub-alpine lakes, such as Lakes Tahoe and Donner (Lowery & Holdich, 1988; Lewis, 2002). However, it also grows well in culture ponds. It is tolerant of brackish water and high temperatures. It does not occur in waters with a pH lower than 6.0. *P. leniusculus* is very active and migrates up and down rivers, as well as moving overland around obstacles. However, their rate of colonization is relatively slow and may only be about 1 km yr-1. Their burrows can reach high densities, i.e.14 m-1, and they can have a serious impact on bank morphology, causing them to collapse. It was considered to be a non-burrowing species, but in Europe in constructs burrows under rocks or in river and lake banks (Guan, 1994; Sibley, 2000).

P. leniusculus is a opportunistic feeder, although more animal than plant material may be consumed if available. It can have a considerable impact on populations of macro-invertebrates, benthic fish, and aquatic plants (Guan & Wiles 1997; Nyström, 1999; Lewis, 2002), it also has been used to clear weed from ponds on fish farms. Griffiths et al. (2004) found that the presence of P. leniusculus significantly reduced the number of Atlantic salmon using shelters in artificial test arenas. Sooty crayfish have become extinct partly due to interspecific competition with P. leniusculus, which was introduced into its range. *P. leniusculus* has also been implicated in causing a reduction in the range of the already narrowly endemic shasta crayfish.

As an opportunistic polytrophic feeder, *P. leniusculus* will eat anything that is available, including other crayfish. The diet was found to shift from aquatic insects in juveniles, to more plant material in adults in some American populations (Lewis, 2002). However, Guan & Wiles (1997) found that cannibalism increased with size and that more animal than plant material was consumed by adults in a British river.

The breeding cycle is typical of a cool temperate zone species, although *P. leniusculus* grows faster and reaches a greater size than its counterparts. Size at maturity is usually 6-9 cm TL at an age of 2-3 years, although maturity can occur as early as 1 year. Mating and egg laying occurs during October in the vast majority of populations. Egg incubation time ranges from 166 to 280 days. In natural populations hatching occurs from late March to the end of July depending on latitude and temperature. Egg numbers usually range from 200 to 400, although some individuals of 66 mm CL have been reported as having over 500 eggs. Based on the use of the lipofuschin technique it has been estimated that some individuals can live 16 years, and other estimates state that it may be as long as 20 years. Some individuals may grow to a large size, i.e. 95 mm CL, but this may not represent a great age, but that of a fast-growing newly introduced population that encounters little competition. Estimates of survivorship to age 2 vary from 10-52%, being dependent on both abiotic and biotic factors. Competition and cannibalism can greatly affect survival in dense populations. Stebbing et al. (2003) demonstrated for the first time the presence of a sex pheromone, released during the breeding season by mature females, that stimulates courtship and mating behavior in male *P. leniusculus*.
P. leniusculus has a typical life cycle of a member of the crayfish family Astacidae, and which is therefore very similar to that of indigenous European crayfish. The eggs hatch into miniature crayfish that stay with the mother for three stages, the third stage gradually becoming more and more independent of the mother. Juveniles undergo as many as 11 moults during their first year, but by age 3 this is reduced to two moults per year, and by age 4 onwards to one moult per year (Lewis, 2002).

Distribution: (Figure 2, North American Distribution)
Native range: Endemic to western North America between the Pacific Ocean and the Rocky Mountains. Occurs from British Columbia in the north, central California in the south, and Utah in the east.
Known introduced range: USA: many states. Europe: Austria, Belgium, Czech Republic, Denmark, England, Finland, France, Germany, Hungary, Italy, Kaliningrad (Russia), Latvia, Lithuania, Luxembourg, Netherlands, Poland, Portugal, Scotland, Spain, Sweden, Switzerland and Wales (Holdich, 2002; Machino & Holdich, 2005; and unpublished information). Japan: Hokkaido (Hiruta, 1996; Kawai & Hirata, 1999), and Honshu (Hiruta, S., 2005, pers. comm.).

Invasion pathways to new locations
Aquaculture: *P. leniusculus* was first introduced into Japan from North America for use as food in 1928 (Kawai et al. 2002b).
Natural dispersal (local): It can disperse along watercourses through natural colonization.

Management information: There are no documented control agents for the successful management of *P. leniusculus* available at this time (Holdich et al. 1999). Trapping is size selective and the smaller individuals remaining take advantage of the lack of competition to grow rapidly (Sibley, 2000). Preventing the further introduction of this species into new bodies of water is one of the few options available. Educating the public to the environmental risks this species pose and identifying new populations are key elements to stopping the spread of this species where it is not wanted. Stebbing et al. (2003, 2004) have researched into the possibilities of using pheromones to attract male *P.leniusculus* into traps. Stringent legislation has been applied to *P. leniusculus* in Britain, which effectively makes it a 'pest' and bans the keeping of it in Scotland and Wales and much of England (Holdich et al. 2004). Despite this P. leniusculus continues to spread and may well cause the extinction of the single indigenous crayfish species within 30 years (Hiley, 2003; Sibley, 2003). Work is in progress in the UK to assess the use of natural pyrethrum again nuisance populations of P. leniusculus in enclosed water bodies (Peay, 2005).

Literature Cited:

Rusty crayfish (*Orconectes rusticus)*

Description: *O. rusticus* can be identified by it's more robust claws and by the dark, rusty spots on each side of their carapace. The spots may not always be present or well developed on rusty crayfish from some waters. In the spring, males will molt into a sexually inactive form (called Form II) and then molt back into the reproductively competent form (Form I) in summer. Form I males are characterized by large claws, a hook on one pair of their legs, and hardened gonapods. The hook and the larger claws are used for grasping females during mating. Males are usually larger than females of the same age.

Ecology: According to Bowen (2003), "*O. rusticus* feed on a variety of aquatic plants, benthic invertebrates (like aquatic worms, snails, leeches, clams, aquatic insects, and crustaceans like side-swimmers and waterfleas), detritus (decaying plants and animals including associated bacteria and fungi), fish eggs, and small fish." *O. rusticus* grow larger, hide less from predators, and attain higher population densities. Therefore they need to feed more. *O. rusticus*, especially juveniles, feed heavily on benthic invertebrates like mayflies, stoneflies, midges, and side-swimmers. It has been estimated that rusty crayfish might consume twice as much food as similar-sized *O. virilis* because of their higher metabolic rate.

According to Bowen (2003), "mature *O. rusticus* mate in late summer, early fall, or early spring. The male transfers sperm to the female, which she then stores until her eggs are ready to fertilize, typically in the spring (late April or May) as water temperatures begin to increase. The stored sperm are released as eggs are expelled and external fertilization occurs. The eggs are then attached to the swimmerets on the underside of the crayfish's abdomen ("tail section"). Just prior to egg laying, white patches appear on the underside of the abdomen ("tail section"), especially on the tail fan. These white patches are glair, a mucus-like substance secreted during egg fertilization and attachment. *O. rusticus* females lay from 80 to 575 eggs. It is important to note that it is not necessary to have both a male and a female crayfish to begin a new infestation. One female carrying viable sperm could begin a new population if released into a suitable environment. Rusty crayfish readily mate in captivity so it is reasonable to expect that mature females, whether used as fishing bait or as science class study specimens, could produce offspring."

According to Bowen (2003), "eggs hatch in three to six weeks, depending on water temperature. Once hatched, young crayfish cling to the female's swimmerets for three to four molts (molting is when crayfish shed their old shell to allow growth). Young crayfish may stay with the female for several weeks. She offers them protection during this vulnerable life stage. Eventually, the young leave the female. They undergo eight to ten molts before they mature, which may occur during the first year, but more likely the following year. Rusty crayfish reach maturity at a total length of one and three-eighths inches and reach a maximum length of about four inches (not including claws). A typical rusty crayfish lives three to four years." A mature adult male molts twice a year and a female molts once hence why males of the same age are usually larger.

According to Bowen (2003), "*O. rusticus* inhabit lakes, ponds, and streams. They prefer areas that offer rocks, logs, or other debris as cover. Bottom types may be clay, silt, sand, gravel, or rock. Rusty crayfish inhabit both pools and fast water areas of streams. They generally do not dig burrows other than small pockets under rocks and other debris, although there have been reports of more substantial burrows. *O. rusticus* need permanent lakes or streams that provide suitable water quality year-round."

According to Bowen (2003), "invading *O. rusticus* frequently displace native crayfish, reduce the amount and kinds of aquatic plants and invertebrates, and reduce some fish populations. O. rusticus is an aggressive species", according to Munjal and Capelli (1982, in Bowen, 2003), "that often displace native or existing crayfish species." According to Bowen (2003), *O. rusticus* displaces native crayfish by crayfish-to-crayfish competition and increased fish predation. The reason for increased fish predation on native crayfish is because *O. rusticus* force the native species from the best daytime hiding places and native crayfish try to swim away from a fish attack instead of taking the more effective claws-up defensive posture the *O. rusticus* does. Perhaps the most serious impact is the destruction of aquatic plant beds that *O. rusticus* causes. *O. rusticus* have been shown to reduce aquatic plant abundance and species diversity which can be especially damaging in areas that are relatively unproductive. These aquatic plants are important for habitat for invertebrates, food for fish and ducks, shelter for young game fish, pinfish, or forage species of fish, nesting substrate for fish, and erosion control (by minimizing waves). Although other crayfish eat aquatic plants, *O. rusticus* eat even more because they have a higher metabolic rate and appetite. *O. rusticus*, especially juveniles, feed heavily on benthic invertebrates like mayflies, stoneflies, midges, and side-swimmers. So, they are more likely to compete with juvenile game fish and forage species for benthic invertebrates than are native crayfish species. Crayfish are eaten by fish, but because of their thick exoskeleton (shell) relative to soft tissue, their food quality is not as high as many of the invertebrates that they replace. Finally, it has been suggested that rusty crayfish harm fish populations by eating fish eggs. While rusty crayfish have been observed to consume fish eggs under various circumstances according to Horns and Magnuson, (1981, in Bowen, 2003), there is no scientific study directly linking fishery declines with crayfish egg predation. It's likely that those fish species that lay eggs in relatively warm water (greater than 50° F) are more susceptible to crayfish predation than fish that spawn during colder water periods. No detailed research has been done that proves rusty crayfish cause declines in fish populations.

<u>Distribution</u>: (Figure 2, North American distribution.)
<u>Native range</u>: Indiana, Ohio, Kentucky, and Michigan in the United States.
<u>Known introduced range</u>: Has invaded many areas surrounding it's native range. It has moved as far west as North and South Dakota, north as Canada and Maine, and south as Tennessee. *O. rusticus* is currently not found in Utah.

<u>Invasion pathways to new locations</u>: Anglers using crayfish as bait are thought to be the primary cause of introduction. Developing a viable commercial harvest of *O. rusticus* from natural lakes could be incentive for trappers to plant them in other waters (Bowen, 2003). According to Bowen (2003), *O. rusticus* are sold to schools by biological supply houses. Even though a warning not to release *O. rusticus* into the wild accompanies crayfish sold to schools, such warnings may be forgotten, or live crayfish may be given away to students and they may eventually be released into the wild.

<u>Management information</u>: Some researchers have suggested that nuisance populations of rusty crayfish are the result of poor fishery management and that by restoring a healthy population of bass and sunfish, *O. rusticus* would be less disruptive in some lakes. The best method of control is to prevent their introduction. Educating anglers, crayfish trappers, bait dealers, and teachers about the threats posed by *O. rusticus* will help reduce the risk of spreading *O. rusticus* to new areas. According to Bowen (2003), "environmentally-sound ways to eradicate or control introduced populations of O. rusticus have not been developed, and none are likely in the near future. The best way to prevent further ecological problems is to prevent or slow their spread into new waters. Regulations in both Minnesota and Wisconsin now make it illegal to introduce *O. rusticus* into any waters. In Minnesota, it is illegal to sell live crayfish as bait and a Department of Natural Resources permit is required to commercially harvest or culture crayfish. Intensive harvest will not eradicate or control crayfish, but may help reduce adult populations and minimize some impacts."
Many chemicals kill crayfish. Some even selectively kill crayfish; however, none are currently registered for crayfish control according to Bills and Marking (1988 in Bowen, 2003). And, none selectively kill *O. rusticus* without killing other crayfish species.

Literature Cited:

Quagga mussel (*Dreissena bugensis*) (N.Muth)

Ecology**:** The quagga mussel is a cousin of the zebra mussel and portrays many of the same characteristics. It is a freshwater, bivalve mollusk that can grow slightly larger than the zebra mussel; up to four centimeters larger. The quagga mussel has a rounded angle, or carina, between the ventral and dorsal surfaces (May and Marsden 1992). The quagga also has a convex ventral side that can sometimes be distinguished by placing cells on their ventral side; a quagga mussel will topple over, whereas a zebra mussel will not (Claudi and Mackie 1994). Color patterns vary widely with black, cream, or white bands. They usually have dark concentric rings on the shell and ventral side and are paler in color near the hinge.

Quagga mussels are filter feeders, removing substantial amounts of phytoplankton and suspended particulate from the water. Impacts on aquatic resources from this filtering are similar to those of zebra mussels. Quagga mussels remove phytoplankton from the water causing alterations in the food web. Impacts associated with the filtration of water include increases in water transparency, decreases in mean chlorophyll, and concentration and accumulation of pseudofeces (Claxton et al., 1998). Increased amounts of pseudofeces in the water have been associated with poor water quality, foul odor and taste. As the waste particles decompose, oxygen is used up, the pH becomes very acidic and toxic byproducts are produced. In addition, quagga and zebra mussels accumulate organic pollutants within their tissues to levels more than 300,000 times greater than concentrations in the environment. These pollutants are found in their pseudofeces, which can be passed up the food chain; therefore, increasing wildlife exposure to organic pollutants (Snyder et al., 1997).

Observations and research suggest the North American quagga mussel is a cold, deep-water form, contrasting with Ukraine populations where the quagga mussel thrives at higher temperatures. In North America, zebra mussels survive indefinitely at 30° C, but the quagga mussel exhibits high mortality at this same temperature (Mills et al., 1996). Although there are indications that quaggas die at lower temperatures than zebra mussels, there are a few exceptional quaggas that are as tolerant of elevated temperatures as zebra mussels, so the potential thermal range of this species may be higher than recent experiments indicate (Mills et al., 1996). Temperature is also a key factor affecting spawning and fertilization in dreissenid mussels. A minimum spawning temperature of 12° C has been reported for zebra mussels compared to 9° C spawning temperature for quagga mussels, which suggests the zebra mussel cannot successfully colonize hypolimnial waters. Although, they have been reported to survive in the hypolimnion, they cannot reproduce there (Claxton and Mackie, 1998). A female quagga mussel with mature gonads was found in Lake Erie at a temperature of 4.8°C, so areas that were thought to be immune to Dreissena colonization may become infested by quagga mussels (Claxton and Mackie, 1998).

Just like zebra mussels, quagga mussels have the capability to attach themselves to any hard surface or substrate; they will even attach on soft substrates and plants. They have the ability to clog pipes used for irrigation, municipal purposes and power generation. Quagga mussels, just like zebra mussels, can cause millions of dollars in damage to our industries. Quagga mussels have a greater tolerance for cooler water temperatures than zebra mussels, and have been found to colonize substrates at greater water depths.

Distribution: Quagga mussels are indigenous to the Dneiper River drainage of Ukraine. It was first documented in the Great Lakes in September 1989, and after confirmation that this mussel was not a variety of zebra mussel, the new species was named "quagga mussel" after the quagga mammal, an extinct African relative of the zebra. Quagga mussels are abundant in the Great Lakes region and more recently have established themselves west of the 100th meridian in the lower Colorado River drainage. In 2007, quagga mussels were confirmed in Lakes Mead, Mojave, and Havasu. Downward drift of planktonic veligers from the aforementioned reservoirs has caused the contamination of the lower Colorado River Basin, areas served by the Central Arizona Project, and areas served by the Southern California aquaduct.

Vectors of Introduction: The rapid invasion and expansion to the west has been exponential due to their ability to disperse at all different stages of life. Quagga mussels move many different ways. The first way they move is naturally, being carried passively as planktonic larvae (veligers) in flowing or wind-driven (wave) water currents and by attaching themselves to other organisms such as crayfish or turtles (Carlton 1994). They may also attach to legs, feet, and feathers of waterfowl and shore birds, but these are only low-level vectors (Johnson 1994). Quagga mussels are mostly transported by humans on their boats. Recreational boating and the ability to move boats and other equipment long distances in short periods of time opens a large introduction capability. All forms of quagga mussels can be transported in many ways including the following: ballast systems, live wells, bait wells, bilge tanks, ski storage areas, cooling systems, and basically anywhere water can be stored. Adult quagga mussels are more likely to attach themselves to boats and equipment and can survive several days out of the water. Some have been known to survive up to 27 days in the right conditions. Quagga mussel veligers are more susceptible to dying in hot, dry conditions. All human forms of introduction can be prevented if the proper precautions and decontamination procedures are followed. Outreach messages across the nation stress “clean,” “drain,” and “dry” all watercraft and equipment having contact with infested waters.

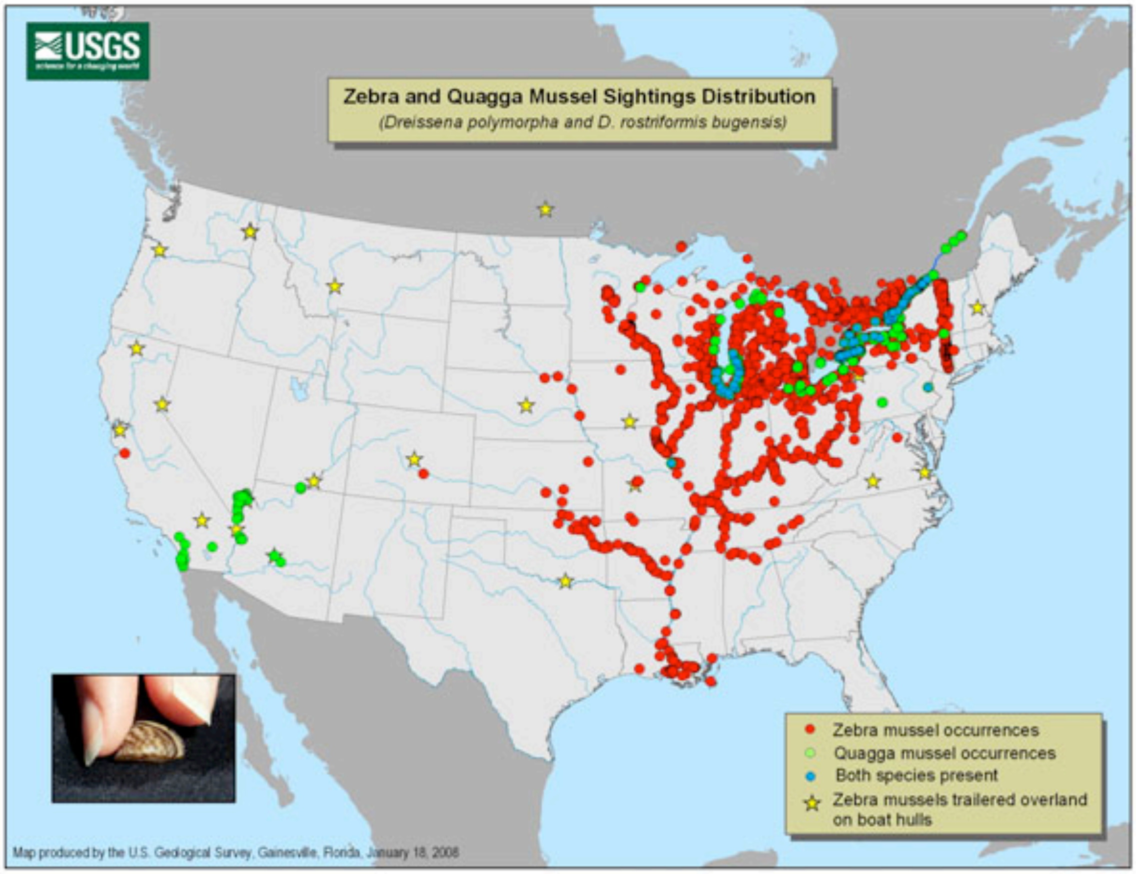
Management consideration: Many different approaches to management have been considered and executed; most resulting in limited or little success. To date, no single “silver bullet” quagga mussel control technology has been identified that will work in all water settings. However, wide arrays of alternative control methods exist for quagga mussels and are suitable or practical for most situations.

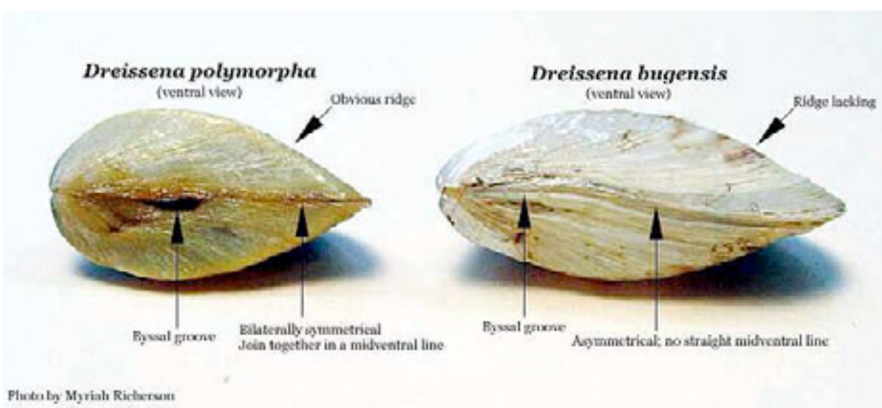
- Manual removal
    - High pressure washer

- Scalding hot water (140º F)
- Manual scraping
- Mechanical filtration
- Disposable substrates
- Molluscicides

- Chemical removal
    - Chlorination
    - Potassium permanganate
    - Metallic salts
    - Non-oxidizing biocides
    - Oxidizing biocides
    - Asphyxiation
    - Thermal treatment
    - Exposure to desiccation
    - Ultraviolet irradiation
    - Biological control
- Prevention of settling
    - High-velocity flow
    - Coatings
    - Electrified surfaces and electrostatic shock
    - Cathodic protection
    - Acoustics
    - Cavitation
- Biological
    - Predators (e.g. freshwater drum, carp, and some sunfish. Also diving-ducks, crayfish and raccoons)
    - Parasites (e.g. trematodes and annelids)

Literature Cited:





Zebra mussel (*Dreissena Polymorpha*) (N.Muth)

Ecology: Zebra mussels are small, freshwater, bivalve (having two matching halves) mollusks with elongated shells typically marked by alternating light and dark bands (zebra stripes). However, shell patterns can vary to the point of having only light or dark colored shells and no stripes. Size ranges very from 1-5mm in their juvenile form to 15+ mm in the adult form. Zebra mussels have byssel threads that allow easy attachment to almost anything. Their considerable genetic and morphological plasticity and broad environmental tolerances enable these organisms to live in a wide variety of habitats. They are prolific, they even attach to each other forming dense layered colonies up to one foot thick. Mussel densities of over 1 million per square meter have been recorded in parts of Lake Erie. Zebra mussels produce microscopic larvae (veligers) that float freely in the water column at numerous depths. Females generally reproduce in their second year by expelling eggs, which are fertilized outside of the body by males: this process usually occurs in the spring and summer, depending on the water temperature. Spawning begins as ambient water temperatures reach approximately 12°C and peaks as temperatures reach the 15°C to 17°C range (Claudi and Mackie 1994). Spawning may be interrupted when temperatures exceed 28°C and will resume when temperatures cool below that threshold (Nichols 1994). Spawning has occurred in the Great Lakes at temperatures as low as 10°C and the larvae have been seen throughout the winter months. Zebra mussel spawning produces planktonic veligers approximately 40ɥm (microns) in length that are capable of active swimming for one to two weeks. Within two to five weeks of hatching, the larval mussels become to large (200ɥm+) and heavy to remain planktonic, and they begin to settle out of the water column (Nichols 1994). At this point mussels, must find a hard substrate to attach themselves to. Once attached, the lifespan of a zebra mussel ranges from 3 to 9 years.

Zebra mussels are diverse, but also have some defined environmental limitations. Zebra mussels can live at water temperatures approaching freezing, but spawning stops below 10ºC and can grow very slowly as temperatures continue to decline. This cold climate can reduce density potentials. Zebra mussels will die when the water temperature falls to levels that would cause ice to form within their bodies. On the opposite end of the temperature spectrum, lethal high temperatures are reached at between 31ºC and 35ºC.

Because zebra mussels need a good deal of calcium to form their shells, requiring water containing more calcium, generally 25 parts per million or greater. Potential for spawning is very low below 9 parts per million of calcium. Zebra mussels thrive in waters that are neither to acidic nor to alkaline, generally pH levels between 7.5 and 8.7. Very low potential exists about 9.0 and below 7.2. The threshold for survival of adults is 6.5 (McCauley and Kott 1993) and for larvae, 6.9 (Mackie and Kilgour 1993). Zebra mussels also require relatively high oxygen concentrations. Little if any colonization will occur at dissolved oxygen concentrations less then 40 to 50 percent full air saturation (McMahon 1995). Velocity of water currents is optimal at 0.09 to 1.0 meters per second. Colonization potential does not become low until velocities either exceed 1.5 meters per second or drop below 0.075 meters per second (O’Neill 1995). Salinity is also a limiting factor in the growth and survival of zebra mussels. Zebra mussels can inhabit brackish areas ranging from 0.2 to as high as 12.0 parts per thousand total salinity (MacNeill 1990).

Zebra mussels adhere to most any surface, including other living organisms in a lake’s ecosystem (e.g. native mussels, crayfish and turtles) Zebra mussels seek out hard surfaces such as: rocks, concrete, steel, pilings, metal grates, boat motors, boat hulls, docks, anchor lines, buoy lines etc. Zebra mussels exhibit some limitations when colonizing which include extensive siltation, microalgea, fluctuating water levels, and antifouling covered surfaces.

Monitoring and control of zebra mussels costs millions of dollars annually and could cost Utah upwards of 15 million dollars a year in maintenance costs (Suflita 2007). Zebra mussels have the biofouling capabilities of colonizing water supply pipes and reducing water flow, inhabiting hydroelectric power plant, disrupting public water supply plants and drastically increasing the maintenance costs at industrial facilities. They are a threat to more than just the world of recreational water use, they are a threat to every person who turns on that tap to get a glass of water, every farmer who uses irrigation pipes or canals where the water is coming from a reservoir, They are a threat to us all here in the west.

Zebra mussels have negative impacts on aquatic ecosystems, reeking havoc on native organisms and native fish populations. Zebra mussels are filter feeders consuming phytoplankton and zooplankton from the water column. Zebra mussels are efficient and can filter up to 1 liter of water per day per individual. They have the capability of filtering an entire lakes volume in a matter of days; it is reported that they filter the entire volume of Lake Erie in 36 hours. This leads to an increase in water clarity and greater penetration of sunlight, allowing development of unwanted macrophytes. The filtering of plankton, which is microscopic, allows the smallest and most basic part of the food chain to be broken, having devastating effects on life cycles of plants, animals, and fish. Zebra mussels can also pollute the water by releasing pseudofeces back into the environment affecting other trophic levels. There are known predators of the zebra mussels such as native birds and some non-native fish, e.g. round goby (*Neogobius melanostomus*), and while the mussel food source may benefit such predators, biomagnifications of toxins into both fish and birds is a potential risk. Loon die offs in recent years on the Great Lakes is strongly suspicisioned to be associated with biomagnification of pollutants due to the loons eating Dreissena mussels.

<u>Distribution</u>: Zebra mussels are native to the Black, Caspian and Azov seas. They were first introduced into North America by transoceanic ships entering the Great Lakes system around the mid 1980's, ultimately being discovered in the United Stated in 1988 in Lake St. Clair. Since this date they have spread throughout the Great Lakes region, along their major tributary and effluent rivers, and they crossed the 100th meridian invading the west in 2007. By late 2007 zebra mussels were known from Pueblo Reservoir in south-central Colorado and San Justo Reservoir in west-central California. They have been interdicted alive on tailored boats, which is the most common method of transportation, in California, Washington, and British Columbia.

<u>Vectors of Introduction</u>: The rapid invasion and expansion to the west has been exponential due ability to disperse at all different stages of life. Zebra mussels move many different ways, the first way is naturally, being carried passively as planktonic larvae (veligers) in flowing or wind-driven (wave) water currents and by attaching themselves to other organisms such as crayfish and turtles (Carlton 1994) They may also attach to legs, feet, and feathers of waterfowl and shore birds, but these are only low-level

vectors (Johnson 1994). Zebra mussels are mostly transported by humans on their boats. Recreational boating and the ability to move boats and other equipment long distances in short periods of time opens a large introduction capability. All forms of zebra mussels can be transported in many ways including the following: ballast systems, live wells, bait wells, bilge areas, ski storage areas, cooling systems and basically anywhere water can be stored on a boat. Adult zebra mussels are more likely to attach themselves to boats and equipment and can survive several days out of the water. Some have been known to survive up to 27 days in the right conditions. Zebra mussel veligers are more susceptible to dying in hot, dry conditions. All human forms of introduction can be prevented if the proper precautions and decontamination procedures are followed. Outreach messages across the nation stress "clean," "drain," and "dry" all watercraft and equipment having contact with infested waters.

Management consideration: Many different approaches to management have been considered and executed, most resulting in limited or little success. To date, no single "silver bullet" zebra mussel control technology has been identified that will work in all water settings. However, a wide array of alternative control methods exists for zebra mussels, and many are suitable or practical for most situations.

- Manual removal
    - High pressure washer
    - Scalding hot water 140º F
    - Manual scraping
    - Mechanical filtration
    - Disposable substrates
- Chemical removal
    - Metallic salts
    - Nonoxidizing biocides
    - Oxidizing biocides
    - Asphyxiation
    - Thermal treatment
    - Exposure to desiccation
    - Ultraviolet irradiation
    - Biological control
- Prevention of settling
    - High-velocity flow
    - Coatings
    - Electrified surfaces and electrostatic shock
    - Cathodic protection
    - Acoustics
    - Cavitation
- Biological
    - Predators (e.g. birds and non-native fish)
    - Parasites (e.g. trematodes and annelids)

Zebra Mussels

Literature Cited:

Conrad's False Mussel (N.Muth)

Vertebrates

Reference Utah Comprehensive Wildlife Conservation Strategy (Wildlife Action Plan) noting that non-native fish species compete with either Tier I (T&E), Tier II (species of onservation concern) or Tier III (species with at-risk habitats) native species and cite (1) Utah Wildlife Code, (2) Collection, Importation & Possession of Zoological Animals and (3) Collection, Importation & Possession of Amphibians & Reptiles as authorities. (intro L.Dalton)

Fish

Mosquito Fish (J.Polloczek)

Burbot (*Lota lota*) (N.Muth)

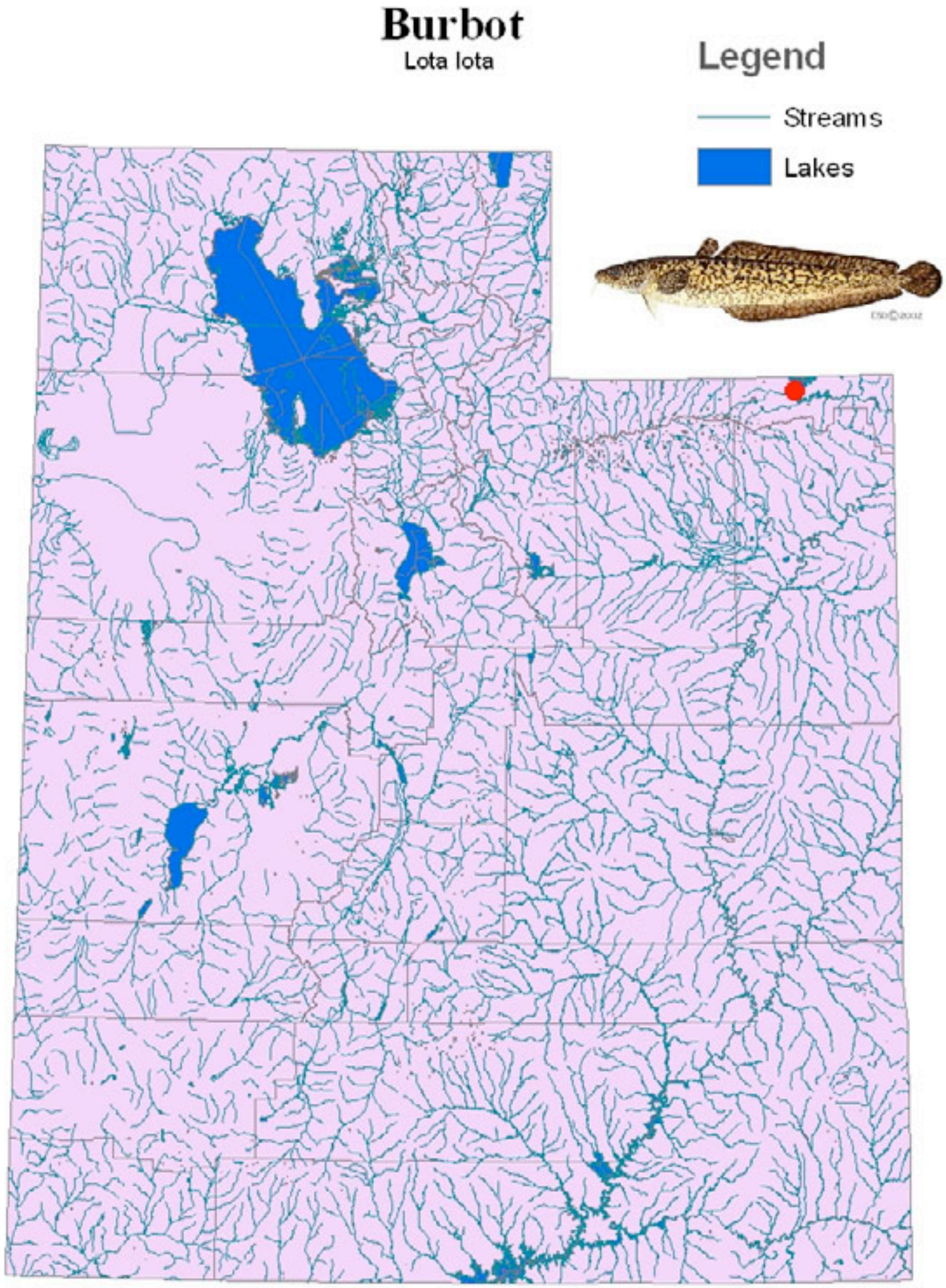
Ecology: Burbot are large fish known to grow to as much as 1.5 meters in length and 34 kilograms in mass (Morrow 1980). These fish are yellow, light tan, or brown with dark brown or black patterning on the body, head, and most fins. The underbelly and pectoral fins are pale to white (Cohen et al. 1990; Morrow 1980). The first dorsal fin is short and is followed by a long second dorsal fin at least six times the length of the first and joined to a rounded caudal fin (Morrow 1980). Burbot have neither dorsal nor anal spines and have 67 to 96 soft dorsal rays, and 58 to 79 soft anal rays (Cohen et al. 1990). Gill rakers are short, pectoral fins are rounded, and caudal fins have 40 rays (Morrow 1980). Like other cods, burbot are also characterized by a single barbel located on the chin (Cohen et al., 1990; Morrow, 1980). Newly hatched burbot are completely planktivorous, and remain so even when they are no longer gape limited (Ghan and Sprules 1993). Diet of larval burbot is dominated by rotifer species for the first two weeks. Diet then shifts to slightly larger nauplii, changing further during week four to cycloid copepods, daphnia, and calanoid copepods (Ghan and Sprules 1993). Juveniles have a diet of molluscs and insect larvae (Tolanen et al. 1999). Adult burbot are piscivorous and consume over 99% fish by mass in Lake Superior (Bailey 1972). Though burbot are always a primarily piscivorous fish, their diet changes seasonally and in response to competition. After the winter months, Tolanen et al. (1999) found that burbot ate a much higher proportion of aquatic invertebrates, namely crustaceans in the early summer and oppossum shrimp in the fall. In the Vilyusk resevoir, their diet overlaps with pike and forces burbot to broaden their diet breadth to include more benthic invertebrates (Kirillov 1988). In addition to fish and invertebrates, Bailey (1972) also found rocks, wood chips, plastic, and other inert materials in burbot stomachs, indicating that burbot feeding habits were somewhat indiscriminate (Bailey, 1972; Ghan and Sprules, 1993; Kirillov, 1988; Tolanen, Kjellmann, and Lappalainen, 1999). Burbot are top predators in their ecosystem, sometimes overlapping with similar top predators such as pike or large salmonids (Kirillov 1988).

Habitat: Burbot are demersal fish found in deep temperate lake bottoms and slow moving cold river bottoms between four and eighteen degrees C (Riede 2004; Cohen et al. 1990). Primarily found at depths ranging from 1 to 700 meters, these fish prefer fresh waters, but are also found in some brackish water systems (Cohen et al. 1990). These fish often dwell among benthic refugia such as roots, trees, rocks, and dense vegetation (Billard 1997). (Billard, 1997; Cohen et al., 1990; Morrow, 1980; Riede, 2004; Scott and Crossman, 1973).

Reproduction & Development: Burbot eggs hatch in the spring between April and June, depending on location (Bjorn 1940; Cohen 1990). Time until hatching is dependent on temperature as well as the particular population and eggs usually take between 30 and 70 days to hatch (MacCrimmon 1959; Bjorn 1940). In four weeks larval burbot increase in length from less than one centimeter to over two centimeter (Ghan and Sprules 1993). Burbot in Lake Superior exhibited very fast growth rates during the first two growing seasons, attaining 42% of total length after ten growing seasons (Bailey 1972). (Bailey, 1972; Bjorn, 1940; Cohen et al., 1990; Ghan and Sprules, 1993; MacCrimmon, 1959). In the Vilyuy River Basin, Siberia, burbot attain sexual maturity in their 7th or 8th year, with males usually maturing one year before females (Kirillov 1988). In Lake Superior, burbot as young as one year old were sexually mature (Bailey 1972). Though sexually mature specimens were found for both sexes in year one and older age classes, there was a higher proportion of sexually mature males until year five when all specimens of both sexes were sexually mature (Bailey 1972). Activity of burbot increases in autumn as energy reserves are concentrated on the growth and development of gonads for the winter spawning season (Kirillov 1988). Maturation of the gonads in both sexes occurs at about four months after the fall peak in nutritional reserves (Pulliainen and Korhonen 1990). (Bailey, 1972; Kirillov, 1988; Pulliainen and Korhonen, 1990).

Burbot breed once per year in the winter, migrating to shallow water or to a smaller stream to spawn (Cohen 1990). Burbot move to spawning areas individually and males tend to arrive before females (Morrow 1980). Spawning occurs during the night when individuals form a globular mass, each fish pushing toward the center and releasing eggs or sperm (MacCrimmon 1959; Cahn 1936). Postspawning runs upstream have been observed, most likely for feeding (Cahn, 1936; Cohen et al., 1990; MacCrimmon, 1959; Morrow, 1980). Burbot are broadcast spawners and provide no parental care. Parental investment in burbot is characterized by an increased metabolic activity level and food consumption rates in the fall in order to contribute to the growth and maturation of gonads in both male and females over a four month period preceeding spawning events (Pulliainen and Kohonen 1990; Kirrilov 1988). It has been suggested that burbot may require one to two years to replenish their nurtritional reserves after each spawning event, but no further information on this topic was available. (Kirillov, 1988; Pulliainen and Korhonen, 1990).

Management Considerations: Burbot are a non-native invasive species probably introduced by sportsman into Flaming Gorge Reservior. Burbot have been found as far south into Utah as Linwood Bay and Antalope Flat. Biologists expect the burbot to move into the canyons and as far south as the Flaming Gorge Dam. The only management tactic that has been tried on the lake, so far, is angling. Burbot have no limit and have a must kill or illegal to release law. Burbot have been caught over the winter months through the ice in large quanities. Because this is a newly introduced species into Flaming Gorge Reservior, DWR, in cooperation with Utah State University, will begin a graduate study in 2008.



Gizzard shad (*Dorosoma cepedianum*) (D.Keller)

Coloration: Back silvery blue, somewhat iridescent; sides silvery above, whitish below; abdomen white. Fins darkened. Dark purplish spot about the size of the eye present immediately behind the upper end of the gill opening in y-o-y. Spot becomes obsolete and disappears with age.

Mouth: small subterminal, slightly overhung by the rounded snout. Centrally notched upper jaw protrudes slightly beyond lower jaw. Maxillary reaching below the anterior margin of the eye. Gill rakers long, slender.

Body: Deep strongly compressed laterally. Average TL 225-350 mm. Scales large, cycloid , deciduous. Lateral line lacking. Median lateral series of scales 61 (52-70). Ridge of sawlike ventral scutes on abdomen.

<u>Ecology</u>: The gizzard shad is common in lakes, oxbows, impoundments, sloughs and large rivers with basic or low gradients (Trautman 1981; Etnier and Starnes 1993), but reaches greatest abundance in waters where fertility and productivity are high (Robison and Buchanan 1988; Pflieger 1997). Gizzard shad avoid high gradient streams and rivers in the mountains and rivers without large, permanent pools, but can tolerate moderately turbid and occasionally even brackish or salt waters (Trautman 1981; Robison and Buchanan 1988; Pflieger 1997). The gizzard shad prefers living in open water, at or near the surface (Becker 1983; Harlan et al. 1987).

The gizzard shad spawns in shallow backwaters or near the shore. Gizzard shad spawn at night, spring to summer, eggs hatch in about 2-4 days. Eggs randomly scatter and adhere to plants, rocks or firm substrate. Spawning may occur when water warms to the high 50's but the peak happens from 66-72 F (19-22 C) during a 6-week spawning period. Fecundity ranges from 22,000 to 350,000. Most spawn at age II during a six-week spawning period. Buoyant larvae become plankton. They reach sexual maturity usually in 2-3 years (Robison and Buchanan 1988). Life span is generally about 4-6 years; few survive beyond age III (Sublette et al. 1990). Typically found traveling in schools, juveniles are nonvisual planktivores, most commonly utilizing zooplankton and phytoplankton in the diet. Adults are primarily bottom filter-feeding detritivores; which typically eat large quantities of organisms attached to underwater surfaces, especially from littoral areas. Gizzard shad also feed on phytoplankton in open water (Sublette et al. 1990).

The gizzard shad feeds by swimming through the water with its mouth open in an apparently aimless manner. Numerous fine gill rakers are present in the gills and act like a very fine sieve; water passes out through the gill slits as the fish swims along, while tiny organism are retained and introduced into its alimentary canal.

Distribution: Gizzard shad were unknown in Utah until 2002 when six individuals were documented in the San Juan arm. They are currently found all over Lake Powell. Since the initial discovery in 2002 Gizzard shad have spread into the Colorado River and Green River systems (fig.1)

In 2006 sampling of the Green River was conducted to evaluate a response of small-bodied native fish to nonnative predator removal. Seining was conducted in suitable low-flow and backwater habitats. Of potential significance in 2006 were the observation of small gizzard shad in backwaters, a decrease in the number of native species, and the number of individuals within each native species. Not all gizzard shad were measured; however, of those that were (n=8), their mean length was 39.75 mm. Lengths of these fish ranged from 36mm to 41mm. Given that fish of such small lengths were found in several backwaters from river mile 281 to 215 (nine total backwaters), the researchers suggested that this species has begun to reproduce in the middle Green River.

Pathways of introduction: It is unknown exactly how gizzard shad where introduced into Utah. It is likely that they came from illegal fish stocking by individuals under the assumption that they would provide good forage for Lake Powell sport fish. Also, they may have been accidentally introduced via fish transport operations from other states in which they are common. It has been reported by U.S. Fish and Wildlife Service that gizzard shad were accidentally introduced into Morgan Lake near Shiprock, NM with a shipment of largemouth bass in 1998. The bass came from Inks Dam National Fish Hatchery in south-central Texas in the Rio Colorado drainage where gizzard shad are abundant in the surface water used at the hatchery. Later loads of bass transported to Morgan Lake from the hatchery were found to have as many as 9 different species besides largemouth bass (fish species included Guadalupe bass, logperch, gizzard shad, white bass, bluegill, and dollar sunfish).

Management considerations: A review by DeVries and Stein (1990) suggests that gizzard shad might not be ideal forage fishes. Gizzard shad can consistently produce large numbers of offspring from few adults (Miller 1960; Pierce 1977), and their larvae may compete with other fishes for zooplankton (DeVries and Stein 1992). Furthermore, because gizzard shad grow quickly (Bodola 1966), they often reach a size refuge from most predators by the end of their first year (Adams and DeAngelis 1987; Johnson et al. 1988). Impressive larval production coupled with fast growth limits predator consumption to a maximum of 30% of gizzard shad production in Ohio reservoirs (Johnson et al. 1988). Most importantly, however, gizzard shad are opportunistic omnivores, feeding on zooplankton as larvae, but capable of switching to phytoplankton or detritus as juveniles and adults (Miller 1960; Bodola 1966; Pierce et al. 1981). As a result, gizzard shad can drive zooplankton to extinction, yet still survive and grow to adulthood. Gizzard shad also spawn before many sport fishes (e.g., bluegill *Lepomis macrochirus*), thus their larvae may deplete zooplankton resources to the extent that sport-fish larvae may face unfavorable conditions for growth and survival.

In 2006 Lake Powell threadfin shad populations decreased as a response to heavy predation from large numbers of adult sport fish, the adult gizzard shad population continued to grow. Due to the suitable habitat available and implications of gizzard shad in Lake Powell, this species will affect the management and planning of recreational sport fishing opportunities of nonnative fish in Glen Canyon NRA. The competitive nature of gizzard shad may pose a threat to endangered species of the Colorado River.

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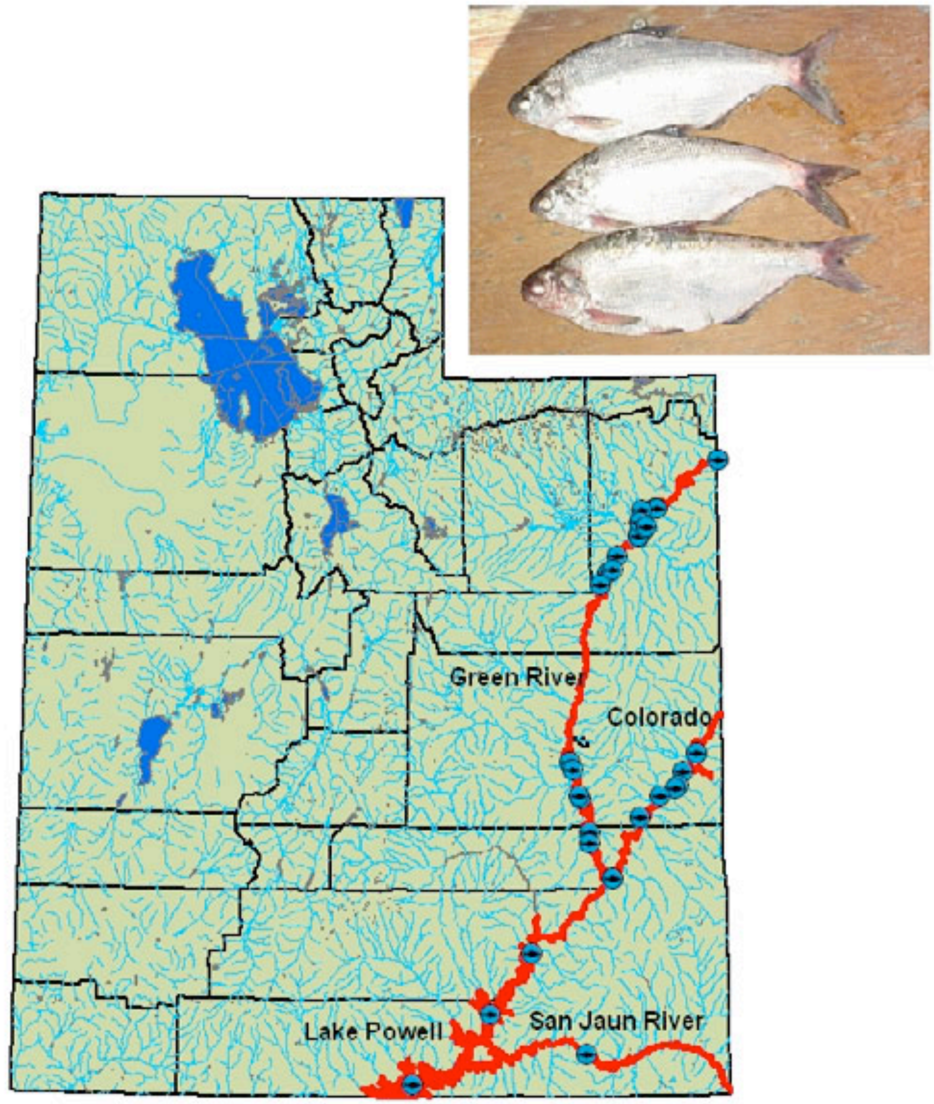
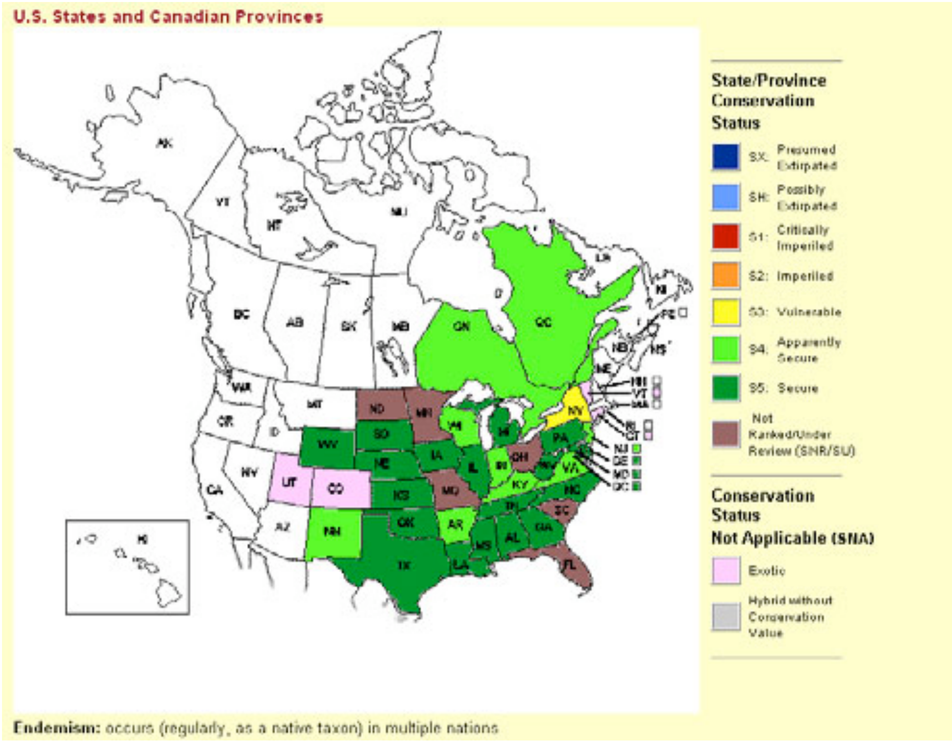


Fig 1. Gizzard Shad (Dorosoma cepedianum) Documented and probable distribution.

Sites where Gizzard Shad have been sampled

Probable Range



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Amphibians

North American Bullfrog (Rana catesbeiana) (N.Muth)

Ecology: North American bullfrogs are the largest true frog found in North America, weighing up to 0.5 kg and 203 mm in length. Typical length ranges from 90 to 152 mm. Color varies from brownish to shades of green, often with spots or blotches of a darker color about the back. The hind feet are fully webbed. The sex of an adult bullfrog can be easily determined by examining the size of the tympanum (the external ear of the frog) relative to that of the eye. The tympanum is a round circle located on the side of the head near the eye, and in males it is much larger than the eye. In females the tympanum is as large or smaller than the eye. Also, during the breeding season the throat of the male bullfrog is yellow, whereas the female's is white. Bullfrogs are normally found in the Eastern US & Canada. They were introduced into California and Colorado in the early 1900's and since then bullfrogs have been introduced in Southern Europe, South America and Asia.

Reproduction: Breeding takes place in May to July in the north, and from February to October in the south. Fertilization is external, with the females depositing as many as 20,000 eggs in a foamy film in quiet, protected waters. Fertilization is usually, but not always, by one male. Tadpoles emerge about four days after fertilization. These tadpoles may remain in the tadpole stage for almost 3 years before transforming into frogs. Adults reach sexual maturity after 3 to 5 years. The average bullfrog lives seven to nine years in the wild. The record lifespan of an animal in captivity is 16 years.
Habitat: North American bullfrogs prefer warm weather and will hibernate during cold weather. A bullfrog may bury itself in mud and construct a small cave-like structure for the winter. Their hunting style is 'sit and wait.' Bullfrogs can wait for a long time for some type of prey to come by, then, pounce on their prey and eat it. Bullfrogs are active both during the day and at night; they are most active when the weather is moist and warm.

Food Habits: Bullfrogs are very aggressive predators. They usually eat snakes, worms, insects, mice, crustaceans, frogs, tadpoles, and aquatic eggs of fish, frogs, insects, or salamanders. They are cannibalistic and will not hesitate to eat their own kind. There have also been a few cases reported of bullfrogs eating bats, and turtles. A good "rule of thumb" for bullfrogs is that if it will fit in their mouths, they will eat it. Bullfrog tadpoles mostly graze on aquatic plants.
Predation: Humans hunt bullfrogs for frog legs, but they have a limited hunting season in most states. Bullfrogs are also eaten by a wide variety of other animals, depending on the region. These include herons, such as great blue herons and great egrets, turtles, water snakes, raccoons, and belted kingfishers. Most fish are averse to eating bullfrog tadpoles because of their undesirable taste. In southern states large mouth bass are their main fish predators.

Management Stratigies: Strategies to control negative impacts from bullfrogs vary from state to state. In California, where predation by bullfrogs on red-legged frogs has been documented, the recommended technique for cattle ponds is draining them entirely while at the same time shooting adults as they attempt to escape (Doubledee et al. 2003). Arizona has employed this technique in numerous isolated areas around the state to benefit various sport fisheries. Colorado allows unlimited statewide harvest of bullfrogs, which can legally be taken by archery, gig, dip net, or by hand. Members of the public still continue to move bullfrogs around in British Columbia, so they have implemented an extensive public education program to increase people's knowledge of the harm that bullfrogs do to native ecosystems. Govindarajulu (2004), after reviewing the situation in British Columbia, concludes that complete eradication is only feasible in small, isolated areas. He does, however, recommend culling metamorphs in the early fall as a method to control their populations (Govindarajulu et al. 2005) vs. removal of adults, which tends to increase populations due to decreased cannibalism. In Utah, along the Wasatch Front, nurseries were giving away bullfrogs with the purchase of backyard water features. Teachers were also receiving bullfrog tadpoles in educational activity kits and then allowing children to take the frogs home when the lesson was completed. In response to these sorts of activities, biologists with the Utah Division of Wildlife worked with nurseries to discontinue giving away bullfrogs. Bullfrogs have been a prohibited species in Utah for quite awhile so it was not difficult to get them to discontinue this activity once they realized it was illegal. Members of the Division also contacted the companies distributing frogs with the educational kits. Educators in Utah will no longer receive bullfrogs if they order from these companies; however, educators in neighboring states can still receive frogs with their order.

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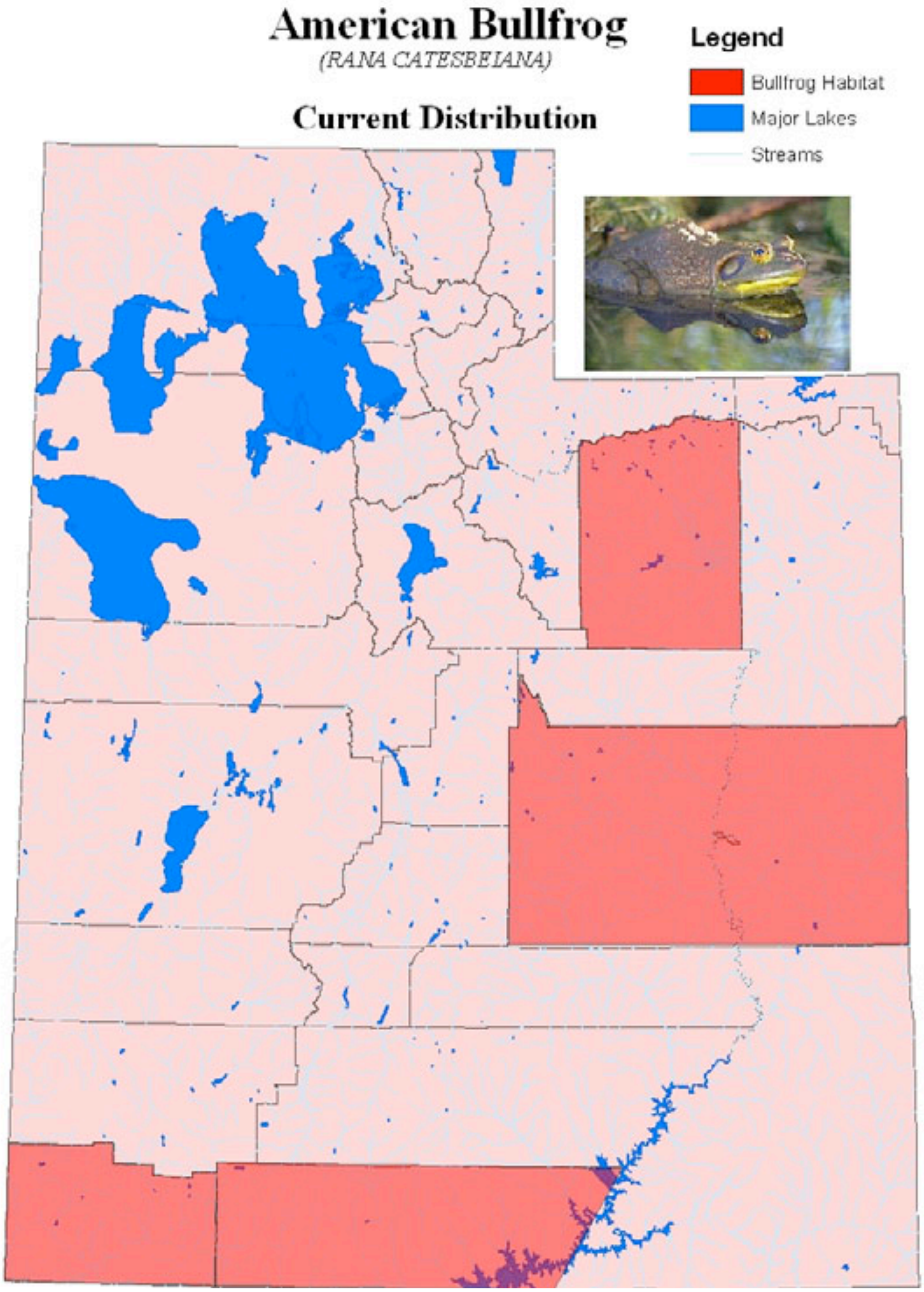


Green Frog (*Rana clamitans*) (E.Freeman)

Ecology: The green frog is a large sized frog with adults ranging in size from two to four inches in length. Life span in the wild is unknown, but captive frogs have been known to live up to ten years. Males and females are phenotypically different. Males have a tympanum that is larger than their eyes as well as having a yellow throat where females have a tympanum that is the same size as their eyes as well as having a white throat. Both sexes have prominent dorsolateral ridges. Both sexes also have dark, transverse bands on their legs as well as well webbed toes. The first fingers do not extend past the second. There are various color phases including bronze, brown, light green and in very rare cases, blue.

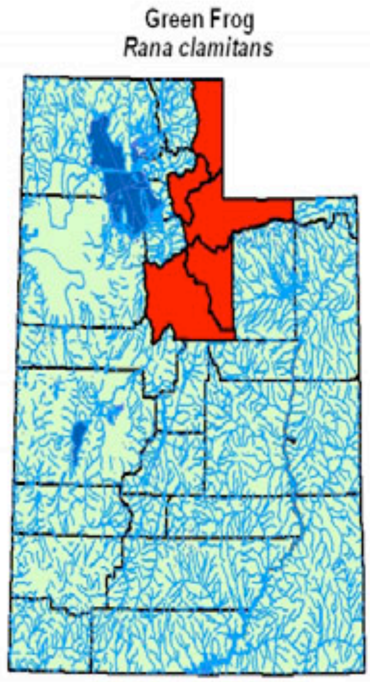
Green Frogs are both diurnal and nocturnal, living and around shallow water. When cold whether months arrive they go dormant until it warms again. Green Frogs are a solitary species except during breeding season when they congregate at breeding locations. Males guard their breeding territory which is approximately one to six meters in diameter and sing to attract females. These frogs also have excellent vision which is used to locate prey. Green Frogs are carnivorous and will eat anything they can get into their mouth. They employ the sit and wait hunting tactic to capture their prey.

Breeding takes place in late spring and lasts between one to three months. Each female produces 1,000 to 7,000 eggs. These eggs are attached to emergent aquatic vegetation or they will float on the surface of the water. Gestation takes three to five days. After hatching the tadpole stage is completed in 3 to 22 months.

<u>Distribution</u>: Green Frogs are native to the eastern United States. They are currently found along the northern Wasatch front in the following Utah counties: Rich, Morgan, Summit, Wasatch and Utah.

<u>Pathways of Introduction</u>: While native to the eastern United States they were likely introduced to the West by way of the pet trade. As their populations grow they will continue to spread throughout the West.

<u>Management Concerns</u>: While not as gregarious as the Bullfrog, the Green Frog does pose a threat to native species. They compete for food and other resources with native fauna, including the threatened Boreal Toad. There are natural predators to these frogs as well as native species including birds and snakes. There are no management efforts that specifically target this species.



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Plains Leopard Frog (*Rana Blairi*) (C.Stock)

Ecology:
The plains leopard frog is about 2.8-3.9 inches long. Its background is brown or green, and has two or three irregular rows of dark spots on the dorsum. This species is often confused with the northern leopard frog (rana pipiens), but it can be distinguished because of a light spot in the middle of the tympanum, a distinct light line along the upper jaw, and dorsolateral ridges that are interrupted just anterior to the groin and medially. It is usually found in streams, reservoirs, ponds, ditches, and other bodies of water.

Breeding occurs in spring and summer. Large egg clusters are attached to submerged vegetation in waters without a strong current.

Distribution: The Plains Leopard Frog is found throughout the Great Plains of the United States, from Indiana west across central and southern plains to South Dakota, south to Colorado, New Mexico, and Texas, with a disjunct population in Arizona. It's current distribution in Utah is the Wahweap area of Lake Powell.

Pathways of Introduction: Most likely introduced by trailered boats into the marina.

Management Considerations: Manual removal can be done at night with a flashlight shined into their eyes. This can be done by gig or by hand. There are also various types of traps that can be set up.

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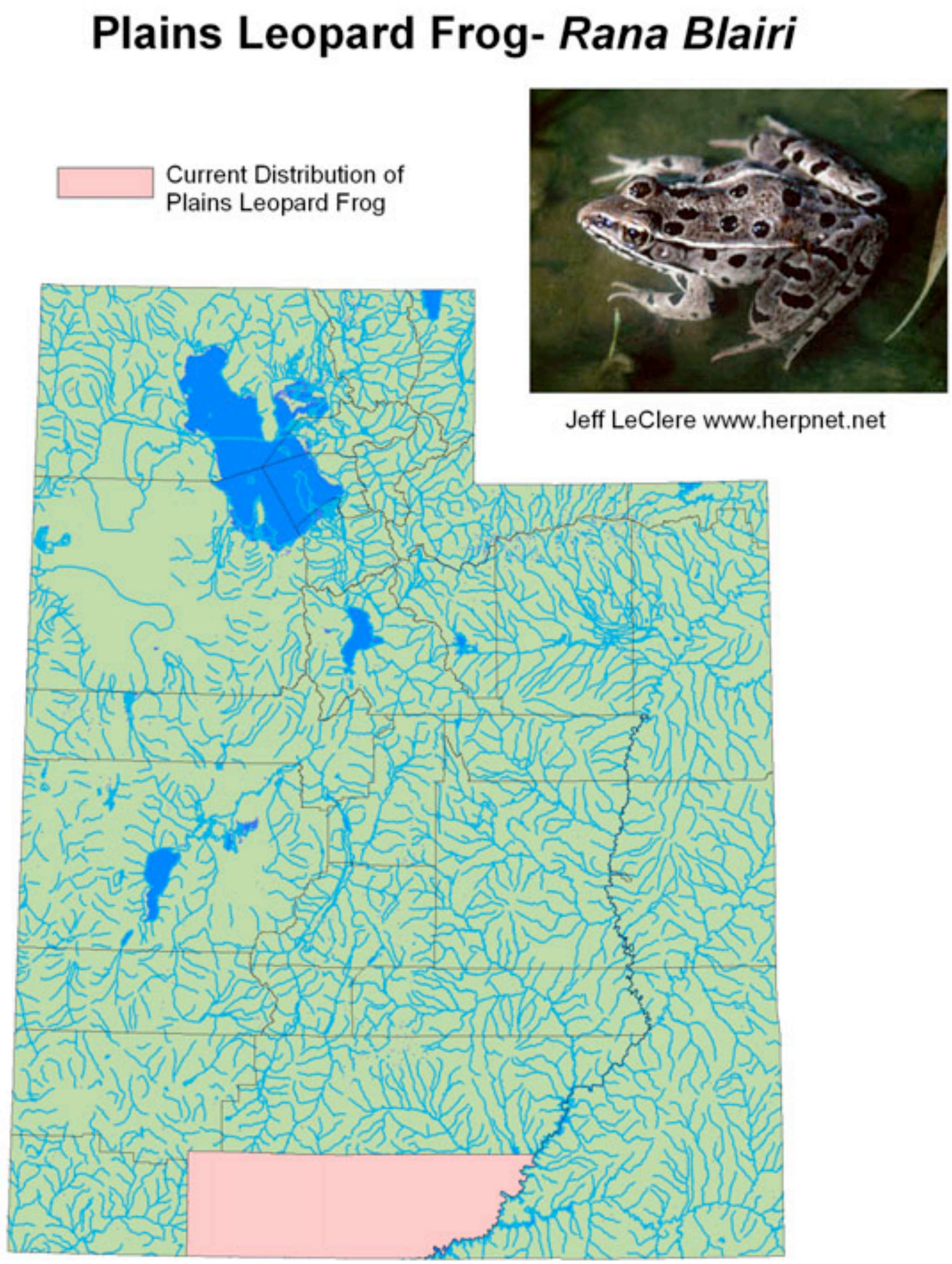

Rio Grande Leopard Frog (*Rana berlandieri*) (C.Stock)

Ecology: The Rio Grande Leopard frog is a highly aquatic frog that is typically found in streams. It is rarely found away from water but can survive by burrowing into moist soils. It is mostly active at night and seldom seen during the day. The diet consists of a wide variety of insects, aquatic prey, and even other frogs. Mating generally occurs after rainfall year round, and generally egg masses are attached to aquatic vegetation.

The coloring pattern is pale green, olive, or a grayish brown. They have dorsal spots that are dark with a light rim, and the thighs have dark reticulations. The frogs also have prominent dorsolateral folds that turn inward in front of the groin. A light stripe also runs along the jaw but fades or completely disappears in front of the eye. Adults are 2.25 – 4.25 inches long from snout to vent.

Distribution: Native to Texas, New Mexico, and Mexico. It is not currently found in Utah, but exists nearby.

Pathways of Introduction: Most likely introduced from trailered boats.

Management considerations: Manual removal can be done at night with a flashlight shined into their eyes. This can be done by gig or by hand. There are also various types of traps that can be utilized.

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Rio Grande Leopard Frog- *Rana Berlandieri*

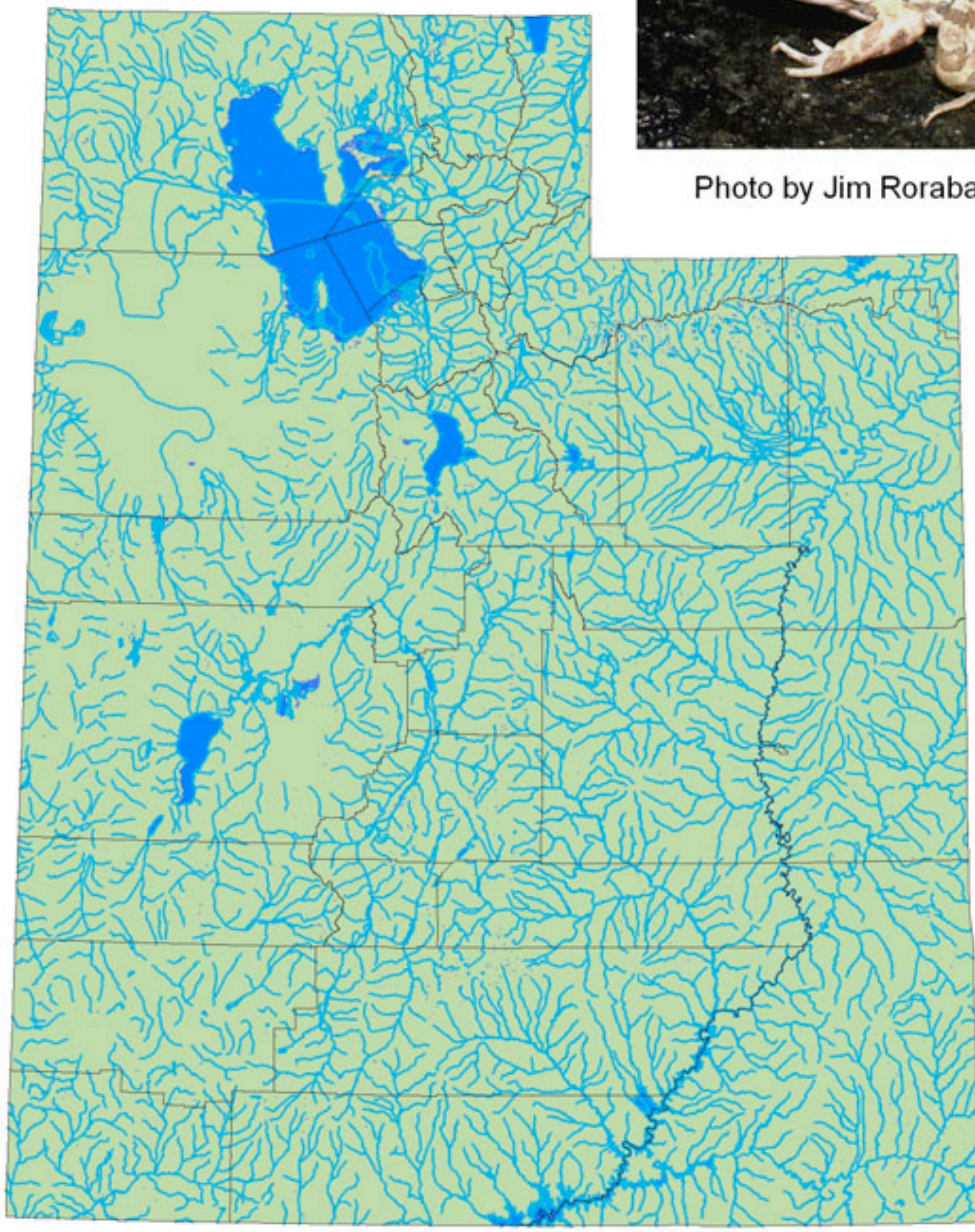

Photo by Jim Rorabaugh

Reptiles

Red-Eared Slider (*Trachemys scripta elegans*) (C.Stock)

Ecology:
Red-eared slides are an aquatic turtle that is commonly sold in the pet trade. The tympanum is covered with red. The back is dark green with black and cream stripes, while the belly has black markings on a cream background. The carapace length of females is 8 inches and males 5-6 inches. Female's shells are domed. The underside of the male's shell is concave, and they have a longer tail than females. Males also have long claws, which is used for mating.

These turtles are often found in fresh and brackish waters, and are a problem because they compete with native aquatic turtles for food. Red-eared sliders are omnivores, and will eat worms, snails, crayfish, small fish, insects, and aquatic plants.

Distribution: Red-eared sliders are native to the Mississippi Valley area of the United States. They currently have established populations in the Washington County and the Weber County areas of Utah.

Pathways of Introduction: Owners release them as they reach adulthood.

Management Considerations: These have become a problem because they are often released into the wild, and they have established populations throughout the United States. Red-eared sliders can be caught using various traps including; floating baited traps, and floating basking traps. Eggs can also be manually removed from females nesting areas. This however must only be done by someone who knows the species very well, and by careful observation.

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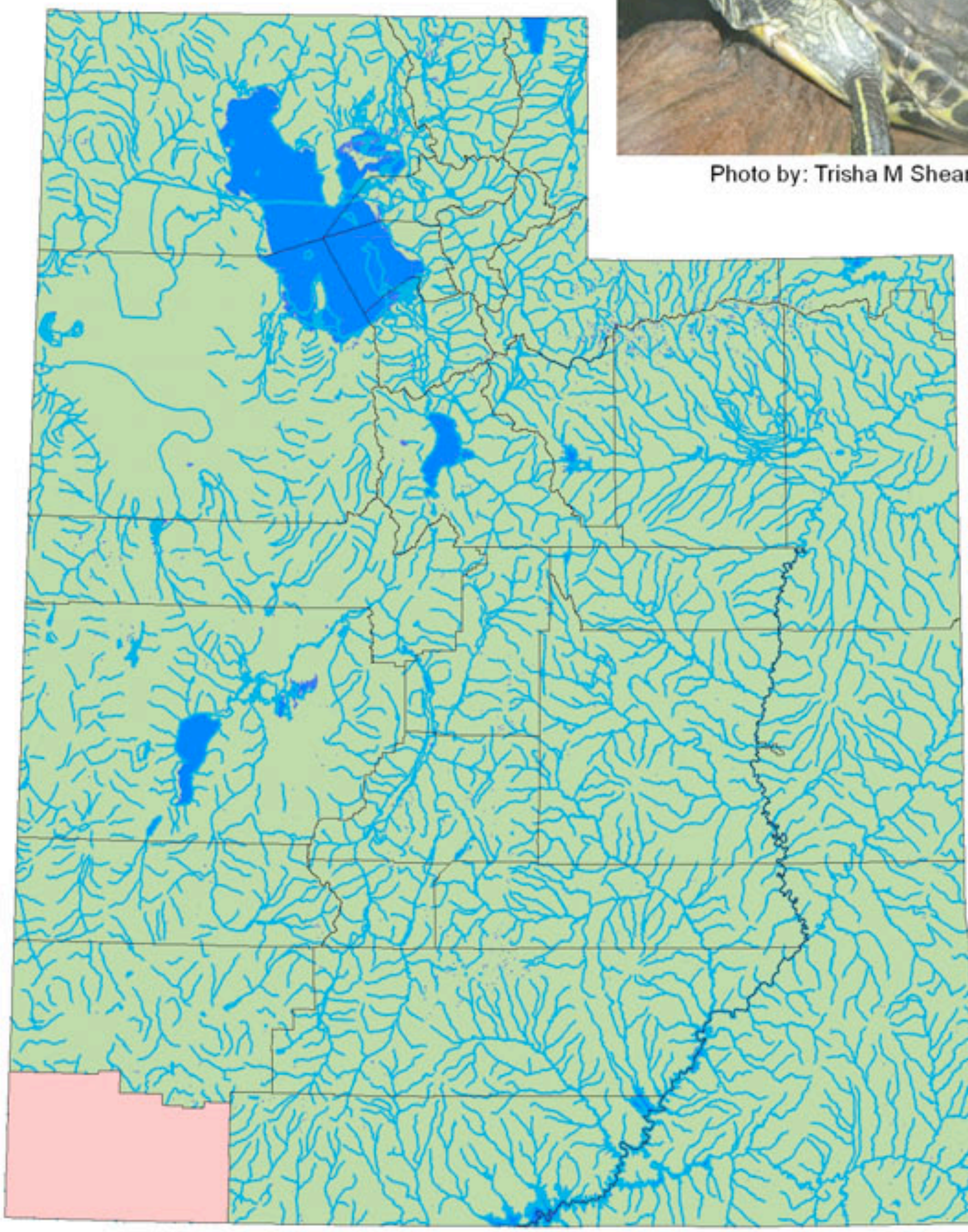
Red-Eared Slider-*Trachemys scripta elega*

Photo by: Trisha M Shears



New Mexico Whiptail (E.Freeman)

Other (Note: Reference other authorities (e.g. Dept Ag ???) (intro L.Dalton)